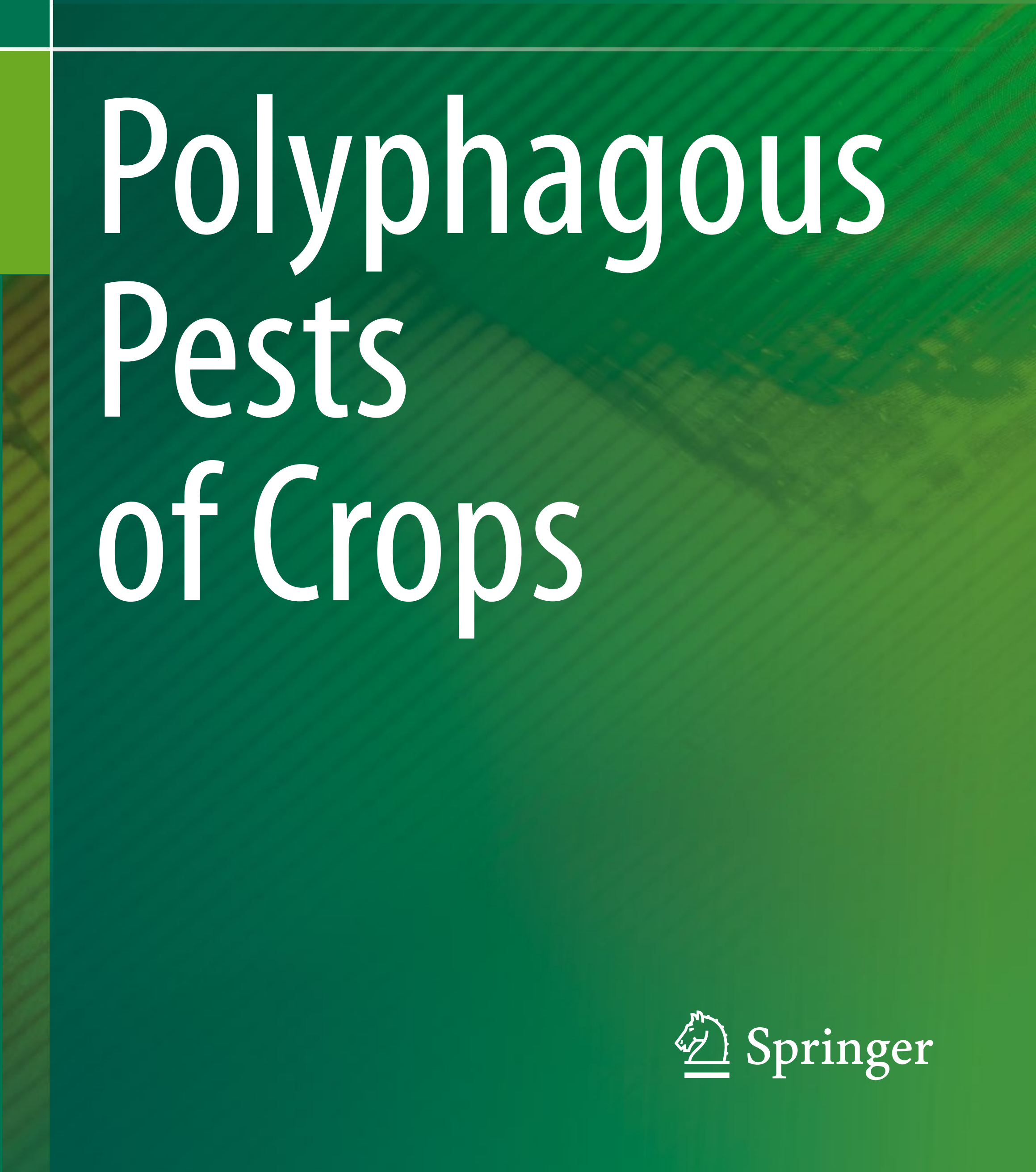
Omkar *Editor*

Polyphagous Pests of Crops



Springer

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Omkar
Department of Zoology
University of Lucknow
Lucknow, Uttar Pradesh, India

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Preface

As human population accelerates and monocultures dominate the agricultural landscape, the reign of insect pests seems to be far from over. Globally, it has been found that the number of insect pests as well as the intensity of their attacks have actually increased post global agricultural revolution. It has been observed that some 137 pathogens and pests cause losses of 10–40% in the staple crops, viz. wheat, maize (corn), soybeans, rice and potatoes, that fulfil about 50% of the calorie intake of humans throughout the world.

While, in general, the losses due to insect pests have increased, monocropping and increased travel of food produce across the globe have especially created a haven for the most dreaded of all insect pests, the polyphagous ones. Polyphagous pests are primarily agricultural pests that feed on economically important agricultural and horticultural crops with wide taxonomic diversity across the globe. They cause massive amount of damage across crop varieties owing to their generalist food habits. The normal cultural practices of crop rotation and plantation of resistant plants find very limited applicability against polyphagous pests.

This book focusses on polyphagous pests not limited to insect pests but also acarine and mammalian (rodent) pests. It covers locusts, termites, aphids, whiteflies, mealybugs, scale insects, gram pod borer, fall armyworm, thrips, mites and rodents. While some of these, such as locusts, termites and rodents, strike a note for both non-specialists and specialists, others have largely vaguer connotations. This book seeks to emphasize how each of these pests is unique in its spread and has the ability to cause extensive damage. Several of these polyphagous pests, in addition to causing direct crop damages, also act as carriers of plant disease-–causing agents. Not only will this book exclusively concentrate on the enormity of losses and the mechanism of losses, but also the means to reduce these losses. In most books dealing with agricultural pests, the emphasis is largely on crop–pest complexes and that too only of insect pests. This book proposes to move beyond specific crops and emphasizes on the pests and view in its entirety and complexity. This unique perspective will provide researchers with better pest management practices that will be more pest specific rather than cross specific and will likely help reduce the cross movement of pests across crops. Not only will this book act as a reference guide to researchers but also to policy planners, academicians and students of entomology.

Right at this moment, globally, polyphagous pests are rising up again. The Middle East countries, the African continent and even the Indian subcontinent are increasingly facing massive locust swarms, which had been under suppression for quite some time. *Helicoverpa* is knocking at the doors of North America, after having extended its range from the traditional old world distribution in southern America. In India, *Helicoverpa* is eating away around 45% of the chickpea crop. The fall armyworm is being feared for its potential spread into new areas. Such resurgences up the ante to study and research more on polyphagous pests; thus, I was motivated to bring together experts on each polyphagous pest and edit a book that would provide the latest information and perspectives on these global pests of crops.

At the very outset, I take this opportunity to express my gratitude to the contributors of different chapters contained in this book for sparing time from their routine to prepare their respective chapters.

I am also thankful to Dr. Geetanjali Mishra, associate professor, Department of Zoology, University of Lucknow, for her intellectual inputs and other final stage support and Ms. Apoorva Shandilya for her assistance throughout this project. I would also like to thank my research students, Dr. Shashwat Singh, Dr. Swati Saxena and M/S Priya Singh, Chandni Verma, Tripti Yadav, Shriza Rai, Lata Verma, Deepali Gupta, and Gauravanvita Singh, for their assistance and support in various ways.

Thanks are also due to my wife, Mrs. Kusum Upadhyay, for her encouragement and support by sparing me from household activities for this work.

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Lucknow, Uttar Pradesh, India Omkar
16 April 2020

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About the Editor

Omkar has been associated with the Department of Zoology, University of Lucknow as a faculty for the past 32 years and a former Head of Department. His specialization is Entomology in which he specifically concentrates on agricultural pests, dealing with the aphids and their natural enemy complex, specifically ladybird beetles. He is author and editor of several books, including Industrial Entomology, Pests and Their Management, and Sucking Pests of Crops, published by Springer Nature. He is an elected Fellow of The National Academy of Sciences, India (FNASc) besides many more.

Contributors

M. Abdollahipour Department of Entomology, Faculty of Agriculture, Tarbiat Modares University, Tehran, Iran

T. Boopathi ICAR- National Bureau of Plant Genetic Resources, New Delhi, India

Mani Chellappan AINPVPM, Department of Agricultural Entomology, College of Horticulture, KAU, Thrissur, Kerala, India

Bhagirath Choudhary South Asia Biotechnology Centre (SABC), New Delhi, India

Sharanabasappa S. Deshmukh Department of Entomology, College of Agriculture, University of Agricultural and Horticultural Sciences, Shivamogga, Karnataka, India

Y. Fathipour Department of Entomology, Faculty of Agriculture, Tarbiat Modares University, Tehran, Iran

Geetanjali Mishra Department of Zoology, University of Lucknow, Lucknow, India

Jagdish Jaba International Crops Research Institute for the Semi-Arid Tropics (ICRISAT), Hyderabad, Telangana, India

C. M. Kalleshwaraswamy Department of Entomology, College of Agriculture, University of Agricultural and Horticultural Sciences, Shivamogga, Karnataka, India

Bhupendra Kumar Department of Zoology, Banaras Hindu University, Varanasi, India

Vinod Kumari Department of Zoology, University of Rajasthan, Jaipur, India

S. Manzari Insect Taxonomy Research Department, Iranian Research Institute of Plant Protection, Agricultural Research, Education and Extension Organization (AREEO), Tehran, Iran

M. Moghaddam Insect Taxonomy Research Department, Iranian Research Institute of Plant Protection (IRIPP), Agricultural Research Education and Extension Organization (AREEO), Tehran, Iran

S. M. Nebapure ICAR- Indian Agricultural Research Institute, New Delhi, India

Omkar Department of Zoology, University of Lucknow, Lucknow, India

B. M. Prasanna International Maize and Wheat Improvement Centre (CIMMYT), Nairobi, Kenya

K. Shankarganesh ICAR- CICR, Regional Station, Coimbatore, India

Abhishek Shukla Department of Entomology, N.M. College of Agriculture, Navsari Agricultural University, Navsari, Gujarat, India

G. Singh Department of Zoology, University of Rajasthan, Jaipur, India

Rajendra Singh Department of Zoology, Deen Dayal Upadhyay Gorakhpur University, Gorakhpur, India

S. Subramanian ICAR- Indian Agricultural Research Institute, New Delhi, India

Y. Yele ICAR-National Institute of Biotic Stress Management, Raipur, India

Locusts

1

N. P. Singh and Vinod Kumari

Contents

Abstract

The term ‘locust’ is given to those species of grasshoppers, which are capable of forming large swarms under certain conditions. They belong to family Acrididae, suborder Caelifera, order Orthoptera and class Insecta. The locust swarms move over wide and distant areas and cause severe damage to cultivated and natural vegetation, where the swarms settle. There are many species of locusts, amongst which desert locust, *Schistocerca gregaria*, is the most damaging pest. The locust problem is not new. Since the beginning of civilisation, they have been a serious threat to agriculture. A substantial increase in locust population, which may cause serious devastation of crops, is known as outbreak. Each locust can exist in two main forms, i.e. solitary phase (phasis solitaria) and gregarious phase (phasis gregaria), which are quite distinct morphologically as well as behaviourally. An

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N. P. Singh · V. Kumari (✉)
Department of Zoology, University of Rajasthan, Jaipur, India
e-mail: vins.khangarot@yahoo.com

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intermediate phase (phasis transiens) also occurs in between two extreme phases. Each locust is a grasshopper, but every grasshopper is not a locust.

Locust breeding and seasonal migration patterns follow no international boundaries; hence, international intervention and cooperation is essential for their control. A locust plague begins, when sufficient rains are there in potential outbreak areas, simultaneously leading to tremendous breeding and increase in locust populations, resulting into upsurge of locust population. Like other pest management, locust control also aims at protecting agricultural productions.

Current information on locust breeding, density and phase is the starting point of all control operations. Chemical control has superseded various other traditional methods for killing, viz. digging trenches, burning, beating and smoking the hopper bands and swarms.

The first international locust conference took place in Rome in 1920. The International Locust Information Network and Anti-locust Research Centre were established in London in 1930. After 1993, the latest swarms of desert locust invaded India on 21 May 2019, which is still continued in major parts of northwest India. The Locust Warning Organization (LWO) and Field Station for Investigation on Locusts (FSIL), established by the Government of India, are active to control the locust swarms and outbreaks. Using certain improved technology for spraying operations, Global Positioning Systems (GPS) leading to new navigational technology, other modern gadgets and mathematical models under integrated pest management (IPM) programme, we will be able to combat the locust problem with international cooperation.

Keywords

Locust · *Schistocerca gregaria* · Upsurge · Outbreak · Plague · Locust control strategies

1.1 Introduction

The term locust is correctly given to a few species of Acrididae family of the suborder Caelifera belonging to the order Orthoptera and class Insecta (Richards and Davies 2013), having antennae less than 30 segments and tympanum located on the lateral side of the first abdominal segment. They are capable of forming large swarms under certain environmental conditions. Thus, the locust is a migratory grasshopper that swarms at regular intervals. The periodicity of locust swarms is mainly governed by complex environmental factors. The locusts are polymorphic species, which exist in three unstable phases, viz. phasis solitaria, phasis gregaria and phasis transiens, differing in the structure, habit and behaviour. The gregarious and solitary phases differ so much from each other that for a long time they were considered as distinct species. The phasis transiens or transitory phase is a series of transitional forms between the solitary and gregarious phases. The transitory phase includes phasis congregans in between solitary and gregarious phase, and in between gregarious and solitary phase is the phasis dissocians. Pradhan (1969) proposed a theory, explaining probable cause of phase transformation and swarm outbreaks in periodic and cyclic manner. According to this theory, drastic climatic conditions of breeding areas of the locusts are solely responsible for the periodic locust outbreaks.

Swarming takes place in gregarious phase, and diapause occurs in eggs of individuals of this phase. The adult individuals of gregarious phase migrate, and migration occurs on sexual maturation. The solitary phase is characterised by the presence of isolated individuals, which exhibit no sign of migration. The eggs of individuals of solitary phase develop without diapause, and sexual maturity is not followed by colour change and migration. In locusts, the actual stimuli, which promote migration, are internal and are associated with the maturation of gonads.

Locusts and grasshoppers are polyphagous in nature, feeding on a variety of vegetation in the form of various crops, grasses, vegetables, etc. They are distributed throughout the world but abundantly occur in tropical and subtropical regions. The locust problem is not new; since the beginning of human civilisation, grasshoppers and locusts have been among the most devastating threats to agriculture. Locusts and grasshoppers are quite different from other pests. In comparison to others, their population can grow to catastrophic levels and can cause a great damage in a very short time. Locusts have been in competition with human beings, since the agricultural practices started by them have been mentioned in holy books, like Bible and Quran. Uvarov (1944) reported death of thousands of people in the Roman colonies of Cyrenaica and territories of Libya, Algeria and Tunisia from famine due to locust plague. The invasion of locusts destroyed 1,67,000 tonnes of grains in Ethiopia in 1958, which could be sufficient for one million people for a year (Steedman 1988).

Locusts and grasshoppers are distributed all over the world, except Antarctica. Their number is found more near the equator, flourishing in subtropical and tropical countries. Their devastating nature has been of much concern to the economic entomologists. Locust outbreaks have been reported in every continent, except Antarctica, which may affect the livelihood of about 10% of the world population (Latchininsky 2010). Though the damage caused by locusts may be nonsignificant for the entire national economy of a country, it may result into 100% loss of the whole season's work for the farmers, adversely affecting the entire family. Amongst the various locust species, desert locust, *Schistocerca gregaria*, is most known for its wide distribution (Fig. 1.1). The desert locust plague occurred in West Africa in 2004 due to unusual heavy rains with favourable environmental conditions for swarming. To handle this outbreak, millions of USD were spent.

The recent desert locust swarms invaded northwest India on 21 May 2019. These desert locust swarms continued to form during December 2019 in west Rajasthan (Phalodi, 2706 N/7222E, and Bikaner, 2543 N/7125E) and in west Gujarat (Palanpur, 2410 N/7226E). A few more late instar hoppers remained active in Bikaner and Pakistan border. Another generation of locusts had breeding in Rann of Kutch, northwest of Bhuj in Gujarat (2312 N/6954E). In some areas, hoppers and solitary adults were also observed in active state (Fig. 1.2).

According to sources received from the Directorate of Agriculture, Government of Rajasthan, previously the desert locusts invaded in 1993 in an approximate area of 310,482 hectares, while in 2005 an area of 16,440 hectares was infested by these

Fig. 1.1 Distribution of *Schistocerca gregaria* (Forskal 1775), desert locust. (Source: CABI datasheet)

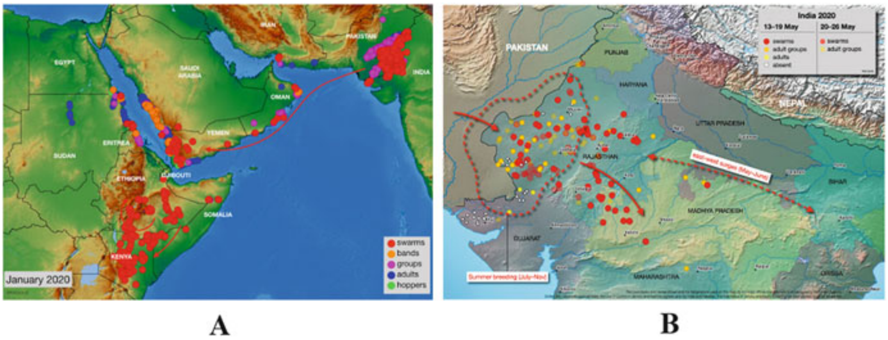


Fig. 1.2 Situation of desert locust. (**a**) Worldwide as on 3 February 2020, (**b**) India as on 27 May 2020. (Source: FAO)

locusts. According to reports of the Food and Agriculture Organization (FAO) of the United Nations, a swarm of locusts comprising of about 40 million individuals can consume the amount of food in a day sufficient for 35,000 people or 20 camels or six elephants. Thus, the destruction to crop yield can be enormous (Fig. 1.3).

Besides being a serious pest of various cultivated and noncultivated crops, locusts (the species is *Locustana pardalina*) have been a delicious food in Africa, Thailand, etc. The tribal people of Uganda and North Central Africa eat *Ruspolia nitidula,* a cone-headed grasshopper. In Kampala (capital of Uganda), the locusts are collected from the street lights. These insects are attracted towards light in huge numbers; when the tribal people collect them, sometimes even the problem of traffic jam is caused.

Fig. 1.3 Hopper bands of desert locust in anonymous field (Rajasthan, India)

Throughout the world, the grasslands and even some forests are permanent home of locusts, and their density varies depending on the season, location and year. The term "outbreak" is used to describe substantial increase in the number of locusts. Outbreak can occur following the arrival of migratory adult locusts. These locusts gather at the site and start breeding (laying eggs) producing enormous individuals. This spectacular congregation results in the formation of hopper bands, subsequently huge swarm of adults, which are capable of migration. Certain advanced agricultural and industrial practices may favour locust outbreaks, such as overgrazing, deforestation, irrigation and introduction of new varieties of crops.

It is estimated that 500 adults of *S. gregaria* per hectare are enough to initiate an outbreak (Fig. 1.4), as they become gregarious. Their colour, morphology, behaviour, biology and ecology are changed. The most significant result of these changes is the formation of hopper bands and swarming adults. Their swarms can migrate hundreds of kilometres per day and invade areas covering millions of square kilometres. The consequences of the swarm invasions can be disastrous for the food production and livelihood of poor rural workers and farmers.

The cost of control practices and strategies is tremendous. The large amount of chemical pesticides affects the environment adversely. This has been a practice for the last so many decades. However, during the past half-century, locust and grasshopper control has made progress through better knowledge of their biology and ecology and increasing the use of high-level technology, such as satellites, high-tech computers and new mathematical models, at our disposal. Previously, due to lack of resources, the information could not be sent well in time by the field workers. Nowadays, special gadgets and wireless sets are provided to the field squads for this purpose, and if needed immediate appropriate action can be taken.

Fig. 1.4 Swarm outbreak in Rajasthan, India (Dec, 2019)

1.2 Major Locust Species

There are around 12,000 locust and grasshopper species throughout the world, of which around 500 species cause damage to crops. The following are particularly important ferocious species of locusts.

1.2.1 The Migratory Locust, *Locusta migratoria* (Linnaeus, 1758) (Fig. 1.5)

The migratory locust (*Locusta migratoria*, Linnaeus, 1758) can go unnoticed for long periods but reproduces continuously, with four or five generations per year. It is distributed in Europe, Africa, Pakistan, Eastern Asia and Australia. Its swarms occurred in Chennai in 1878. This is normally confined to the Central Highlands of Queensland, though low numbers are common as far south as northern New South Wales. There was heavy and concentrated breeding and swarm formation of this locust during February–March 1954 in the Ramanathapuram district of Tamil Nadu, and this was attributed to the swarm observed over Bengaluru (then Bangalore) in June 1954. In general, the locust breeds in Balochistan (Pakistan) during the spring, and the resultant adults migrate to the desert areas of India and breed there during summer. Hoppers pass through five to six instars. In sub-Saharan Africa and Madagascar, four fifths of the islands were affected between 1997 and 2000. The

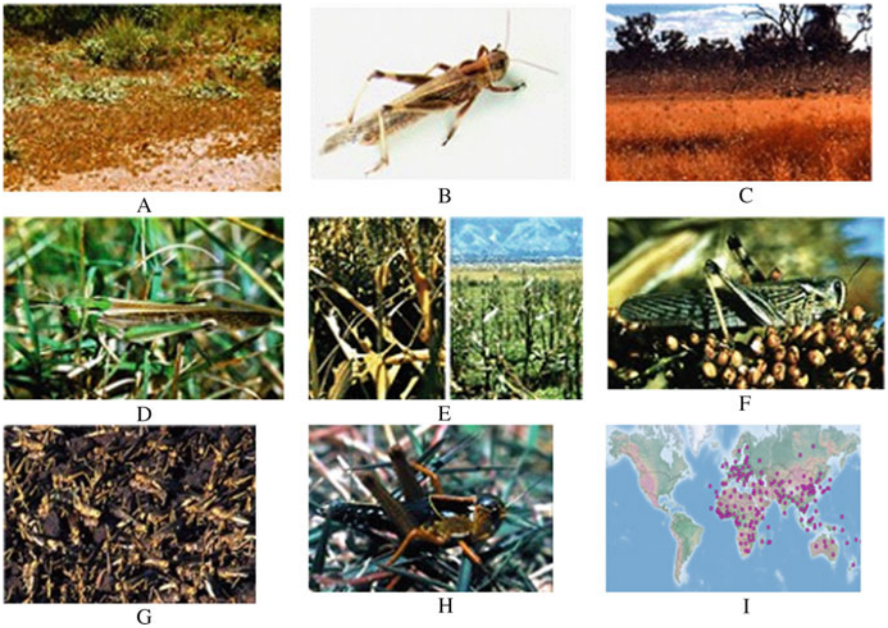


Fig. 1.5 Different stages and distribution of *Locusta migratoria* (Linnaeus, 1758): (**a**) Hopper band, (**b**) adult, (**c**) swarm, (**d**) adult female solitary, (**e**) Left: loose swarm causing damage to maize, Markham Valley, Papua New Guinea. Right: feeding damage on sorghum, Australia, (**f**) adult Female gregarious feeding on sorghum. Females 50–75 mm in length, (**g**) gregarious adults of on egg bed, (**h**) gregarious, fourth instar, length 25–35 mm, (**i**) distribution. (Source: CABI datasheet)

main migratory locust gregarisation area in Africa (where the insect switches from a solitary to a gregarious phase and the first swarms form) is in Mali, on the Niger River floodplain. The last major invasion in this area was in 1928. Within 5 or 6 years, it had spread to a large part of Africa, south of Sahara. It lasted until 1940. In Madagascar, the gregarisation area is in the southwest, the driest part of the island. Invasions starting from there have been known to cover up to 500,000 square kilometres. A large-scale invasion began in Madagascar in 1997. There were also severe outbreaks in the Lake Chad basin in 1997 and on numerous Indonesian islands, particularly Sumatra in 1998.

The hopper bands and swarms are known to attack millet, maize, rice, sugarcane, wheat, banana, pineapple, oil, date or coconut palm, cotton, groundnut, pasture and kitchen garden and even ornamental crops.

Migratory locusts have four generations per year, but egg mortality is reported in dry periods. At high population densities, hopper band and adults form swarm. The solitary form is green or brown, but gregarious (swarming) is straw coloured. Hindwings are greenish yellow without markings. The mandibles are dark purple to black. Hopper bands are generally black and tan coloured. There are many subspecies of migratory locusts with variation in their morphometrics and distributions. Vijay Veer et al. (2013) reported that locust outbreak which occurred in Ladakh during 2006 belonged to subspecies *L. migratoria migratoria*, revealed through molecular characterisation. The following subspecies are described by Mani (1973):

I. *L. migratoria migratoria*—found in southeast Russia
II. *L. migratoria rossia*—found in central Russia and western Europe
III. *L. migratoria capito*—found in Madagascar
IV. *L. migratoria migratorioides*—found in Africa, western Asia
V. *L. migratoria manilensis*—found in Malaysia, East Indies, Philippines and China

1.2.2 The Italian Locust *Calliptamus italicus* (Linnaeus, 1758) (Fig. 1.6)

It is dark brown, yellowish brown or greyish in colour. At the base, the hindwings are rose coloured posteriorly. Forewings project beyond the tip of hind femora with two or three rudimentary brown spots. This species is found in whole of central, meridional and eastern Europe, Caucasia and west Siberia. Breeding takes place in sagebrush steppes and in loose dry soil, with sparse vegetation.

1.2.3 Moroccan Locust, *Dociostaurus maroccanus* (Thunberg, 1815) (Fig. 1.7)

It is testaceous brown, with black spots. Pronotum is with two straight diverging black bands; hind femora is black apically with three black transverse bands, and distal half of hind tibial spines are black. Tegmina are variegated brown; hindwings are transparent. Cerci are short, and lamina is triangular; female is 20–33 mm long, and male 1is 7–28 mm long. This species occurs in the whole of central Europe,

Fig. 1.6 The Italian locust *Calliptamus italicus* (Linnaeus, 1758): (**a**) Male; (**b**) female

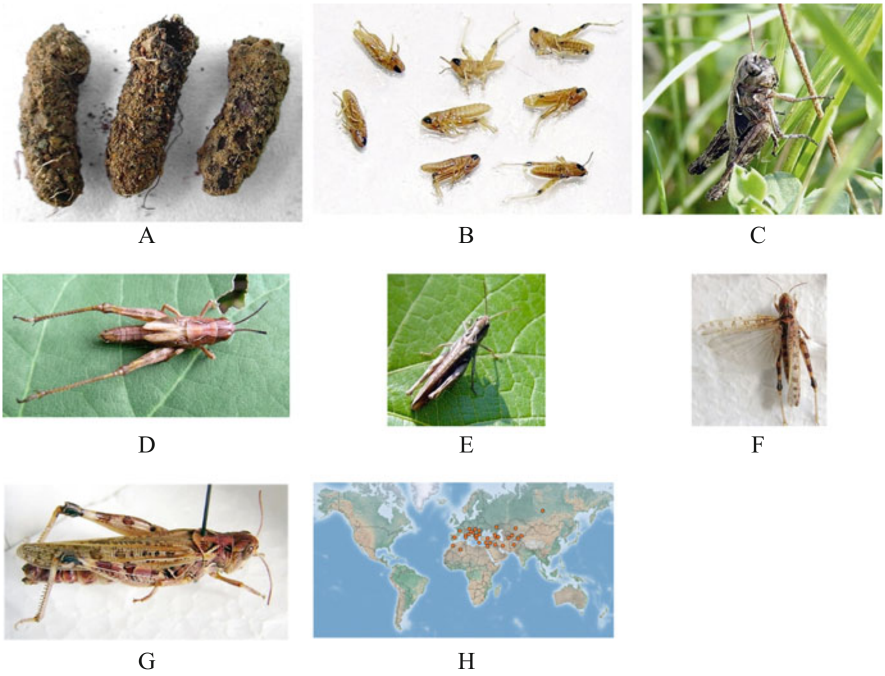


Fig. 1.7 Different stages and distribution of *Dociostaurus maroccanus* (Thunberg, 1815). (**a**) Egg pods. The eggs are whitish to pale yellow, elongated, stick-shaped and arranged in three to four rows in egg pods (oothecae), (**b**) newly hatched nymph, (**c**) third instar nymph on wild grasses. The third instar nymphs have visible wing buds. They are olive red with black spots on the head and body, (**d**) fourth instar nymph on a leaf (note developing wing pads), (**e**) male on vine leaf, (**f**) male with stretched wings; set specimen, (**g**) female set specimen, (**h**) distribution. (Source: CABI datasheet)

Africa from north of Morocco to Tunisia, Asia Minor, Iraq and Transcaucasia. It is specially abundant in Spain. It breeds in hilly or mountainous regions.

1.2.4 Australian Plague Locust, *Chortoicetes terminifera* (Walker, 1870) (Fig. 1.8)

The adults show variation in length ranging from 20 to 45 mm, and colour is grey, brown or occasionally green—often with a pale stripe down the middle of the back. The head is higher than the thorax. The thorax possesses ‘X’-shaped marking. Wings are clear with a dark spot at the end. This is the most economically important Australian locust because of the extent of damage and frequency of outbreaks. It occurs throughout Australia, except moist and cooler regions in the southeast and most of Tasmania.

Adults make short flights just above the grass, often landing side onto the observer. This flight is also typical of several grasshopper species, including the eastern plague grasshopper. This insect looks very similar to *Chortoicetes*

Fig. 1.8 Adult ([A] side view and [B] stretched wings) of *Chortoicetes terminifera* (Walker, 1870)

terminifera, but its hindwings are pale yellow with a dark band, and it lacks the red shanks on the hindlegs.

1.2.5 The Red Locust, *Nomadacris septemfasciata* (Audinet-Serville, 1838) (Fig. 1.9)

Generally, the colour is brown but deepening and reddening with age. Tegmina are with seven oblique, dark bands. This species is distributed in most of South Africa, south of Sahara, Madagascar and Mauritius.

1.2.6 South American Locust, *Schistocerca paranensis* (H. Burmeister, 1861) (Fig. 1.10)

North American locust extends from Argentina north of 40th latitude through Brazil to Venezuela and Central America, but swarms often penetrate Mexico and West Indies. *Schistocerca americana* is probably the solitary phase of this species. The gregarious phase is greyish white, with large black spots on the tegmina, over 50 mm long. It breeds in spring in damp soil.

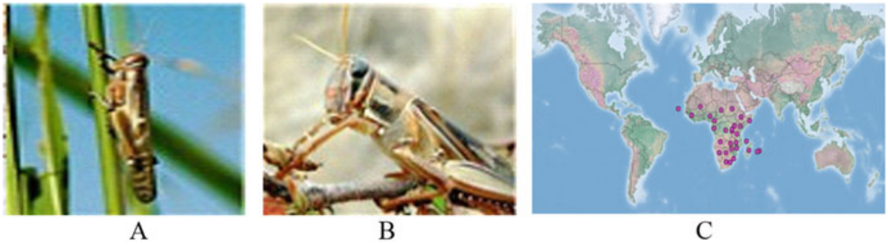


Fig. 1.9 Adult and distribution of *Nomadacris septemfasciata* (Audinet-Serville, 1838). (**a**) Adult on wild sorghum in the Wembere Plains in Central Tanzania in February 2003 (Released into the Public Domain by Christiaan Kooyman), (**b**) close-up of anterior region, (**c**) distribution. (Source: CABI datasheet)

Fig. 1.10 *Schistocerca paranensis* (H. Burmeister, 1861)

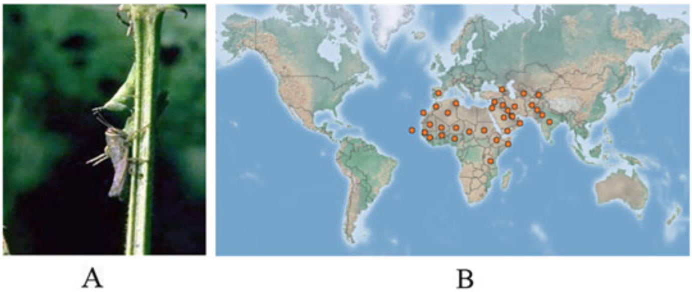


Fig. 1.11 Adult on bean (**a**) and distribution. (Source: CABI datasheet) (**b**) of *Oedaleus senegalensis* (Krauss, 1877)

1.2.7 Senegalese Grasshopper, *Oedaleus senegalensis* (Krauss, 1877)/*O. abruptus* (Thunberg, 1815) (Fig. 1.11)

It is a medium-sized grasshopper species. Though it is not called a locust, this species shows gregarious behaviour, and some morphological changes are like locust on crowding. It is distributed in Sahel region of Africa, the Canary Island and West Asia.

1.2.8 Bombay Locust, *Patanga* (*Cyrtacanthacris*) *succincta* (Linnaeus 1763) (Fig. 1.12)

It is greyish brown, taking on a reddish tinge and darkening with age. There is a broad median pale yellow band on the head and pronotum, continuing down the tegmina. It is distributed in India, Sri Lanka and Malaysia. In India, it is generally restricted to the area extending from Gujarat to Tamil Nadu. It breeds in the Western Ghats and has one generation per year. Hoppers are found scattered among crops or grasses and do not congregate. Its plagues occurred during 1835–1845, 1864–1866 and 1901–1908, and heavy damage was caused to crops. Hoppers of this species pass through seven to nine instars.

Fig. 1.12 : Bombay locust, *Patanga succincta* (Linnaeus, 1763). (Source: Usmani and Usmani 2018)

Fig. 1.13 Egyptian locust, *Anacridium aegyptium* (Linnaeus, 1764) Libyan Desert. (Source: Usmani and Usmani 2018)

Fig. 1.14 Bird locust, *Ornithacris turbida* (Walker, 1890). (Source: Usmani and Usmani 2018)

1.2.9 Egyptian Locust, *Anacridium aegyptium* (Linnaeus 1764) (Fig. 1.13)

It is brownish grey in colour, hindwings are often pale violet at base, and hind tibial spines are pale and dark at the apex. It is distributed throughout Mediterranean region, West Pakistan, Portugal, Spain, France, Italy, Switzerland, Afghanistan, Iran, Iraq, Israel, Jordan, Lebanon, Libya, Egypt and South Arabia. It is generally a minor pest but becomes a major pest when population is built up.

1.2.10 Bird Locust, *Ornithacris turbida* (Walker 1890) (Fig. 1.14)

Generally, bird locusts are similar externally to *Nomadacris septemfasciata*; however, they can be separated by the presence of a triangular side stripe on pronotum which is parallel in *N. septemfasciata*, and hindwings are orange at the base. They are distributed in Libya, Sudan, Uganda, Tanzania, Zaire, Ghana, Togo, Nigeria, Cameron and Angola.

Fig. 1.15 Brown locust, *Locustana pardalina* (Walker, 1870). (Source: Usmani and Usmani 2018)

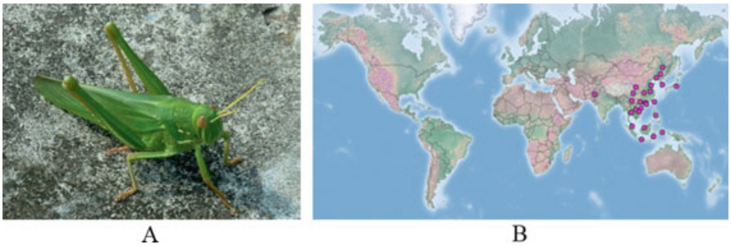


Fig. 1.16 (**a**) Cotton or citrus locust, *Chondracris rosea* (De Geer, 1773). (Source: Usmani and Usmani), (**b**) distribution of cotton or citrus locust, *Chondracris rosea* (De Geer 1773). (Source: CABI datasheet)

1.2.11 Brown Locust, *Locustana pardalina* (Walker 1870) (Fig. 1.15)

Generally, they are brown and rarely green, and tegmina are with scattered brown spots. They are distributed in Zambia, Angola, South Africa, Zimbabwe, Botswana, etc. It is a serious pest in South Africa.

1.2.12 Cotton or Citrus Locust, *Chondracris rosea* (De Geer 1773) (Fig. 1.16)

Wings are tinged green and basally rose. They are distributed in Korea, Japan, Taiwan, China, Hong Kong, Philippines, Thailand, Myanmar, Malaysia, Bangladesh and India. There are many natural enemies of this locust.

1.2.13 Large Coast Locust, *Valanga irregularis* (Walker 1870) (Fig. 1.17)

This is the largest Australian grasshopper species; it is dark greyish brown in colour and with irregular black marks and sometimes with a pale median dorsal stripes. Tibial spines are dark red, and hindwings are with black veins. It is distributed in Australia (New South Wales to Sydney, Queensland, etc.).

1.2.14 The Mediterranean Locust, *Calliptamus barbarus* (Costa, 1836)

It is distinguished from the Italian locust by the single large black spot on the medial side of hind femora. This species occurs abundantly in east Italian and neighbouring regions and extends up to Iran in the east.

1.2.15 The Rocky Mountain Locust, *Melanoplus erectus*

The adults are yellowish brown in colour, with minute dark spots on wings, and are about 35–37 mm. They are distributed in prairies of the eastern slopes of the Rocky Mountains in the USA.

1.2.16 Spur-Throated Locust *Austracris guttulosa* (Walker, 1870) (Fig. 1.18)

This locust is an Australian species. The duration of its life cycle is 1 year. It cannot survive in dried or arid conditions; hence, population buildup of this species takes place during monsoon only. Due to this reason, its plagues are not so frequent as compared to Australian plague locust. However, it may migrate to cultivated fields; if it rains during summer, it may cause severe damage, particularly to seedling stage of sorghum. It may form swarms during overwintered roosting.

The eggs are laid scattered in the fields, not in egg pods. No band formation by hoppers takes place after hatching; hence, the control becomes a bit difficult due to scattered status of nymphs. Hatching takes place from November to February. The overwintered forms become mature during spring, and further egg laying takes place after rainy season in October–November.

The nymphs and adults both possess a peculiar spur between the front legs, hence the name ‘spur-throated locust’. Initially, the colour of nymph is green, but soon a black stripe is developed down the middle of the back. On maturity, the body colour

Fig. 1.17 Large coast locust, *Valanga irregularis* (Walker, 1870)

Fig. 1.18 Spur-throated locust, *Austracris guttulosa* (Walker, 1870)

Fig. 1.19 Yellow-winged locust, *Gastrimargus musicus* (Fabricius, 1775)

becomes light brown. The adults are 50–80 mm in length with pale brown body and longitudinal white stripe. Hindwings are colourless with a blue tinge, and the hindlegs are provided with two rows of white spines with dark tips.

1.2.17 Yellow-Winged Locust *Gastrimargus musicus* (Fabricius, 1775) (Fig. 1.19)

It is a common grasshopper in Australia. It displays its yellow back wings during flight only, when it also clicks with loud clicking sound. It is found in all mainland states. The population is found at its maximum from spring to autumn causing severe damage. At higher population density, the nymphs (hoppers) form bands and the adults swarm. The females lay eggs in the soil in dense egg beds. At normal temperature in summer, which is 28 to 33 °C, its development duration is minimum.

1.2.18 Desert Locust, *Schistocerca gregaria* (Forskal, 1775) (Fig. 1.20)

Schistocerca gregaria is the most widely distributed locust and destructive species of locusts and hence deserves more detailed account. It occurs in a great part of North Africa, southwest (excluding the extreme part of meridional) and most of tropical Africa, Arabia, Iraq, Iran, Afghanistan and northwest India. The heavy swarms may penetrate Spain, Portugal and also the eastern part of India. The distributional range is distinguished into a permanent breeding station and a temporary station into which the gregarious phase migrates. The gregarious form of this species was formerly described as *S. peregrine*. There are generally two annual

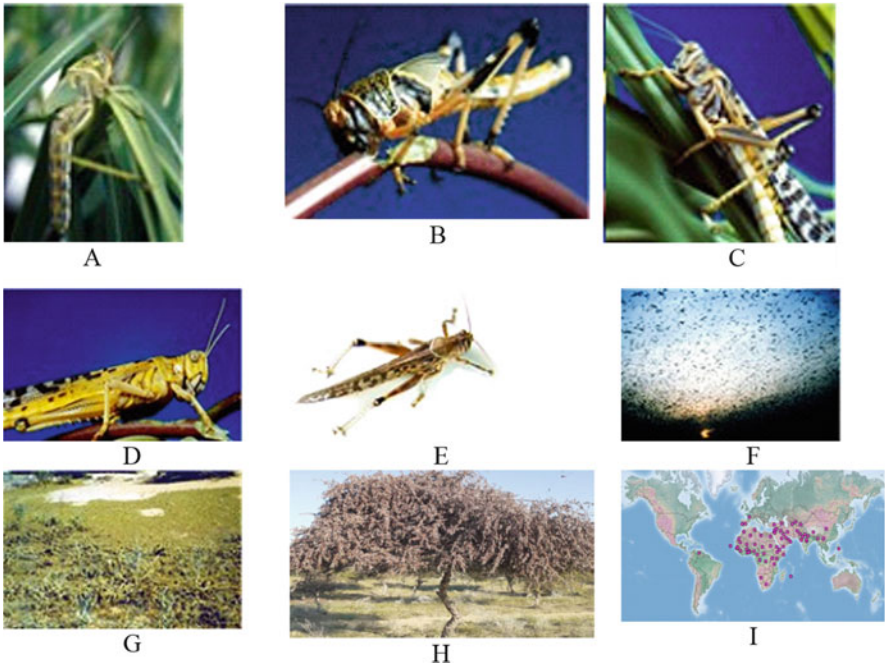


Fig. 1.20 (**a**) Fifth instar nymph of solitary *S. gregaria* (Forskal, 1775), (**b**) fifth instar nymph gregarious, (**c**) adult solitary, (**d**) adult gregarious (Bilkent University, Ankara, Turkey), (**e**) adult (museum set specimen), (**f**) swarm in flight, (**g**) hopper band, (**h**) swarms (adult) on *Acacia* tree, Rajasthan, India, (**i**) worldwide distribution. (Source: CABI datasheet)

generations. The egg stage may last for 15–40 days, and the nymphal (hopper) duration may be comprised of 40–60 days. They breed in permanent breeding grounds, but breeding may be limited due to natural enemies depending on the meteorological or environmental conditions. Essentially, it is hot desert species and is the most destructive. Its ravages have been discussed in biblical times.

The desert locust easily shifts from a harmless solitary form to a damaging gregarious phase. Its ability to move, the high density of its swarms and its voracious appetite are a serious threat to agriculture in many countries. The desert locust habitat covers Africa, north of the equator, the Middle East, the Arab and Indo-Pakistan peninsulas and, sometimes, Mediterranean Europe. It represents 57 countries in all and covers over 20% of the world land area. Between two successive invasions, the locusts retreat to the most arid areas of their usual habitat, where they can go unnoticed for several years. The desert locust does not spare a single crop. It also severely damages the natural vegetation, which has a serious impact on livestock production, as the animals are unable to find enough food. It can also cause havoc on wheat, barley, vines, citrus crops, date palms, millet, sorghum, vegetables, etc., in which countries rely on for food.

1.3 Life History of Desert Locust

Like all other locusts and grasshoppers, the life stages in life cycle of the desert locust are egg, nymph (hopper) and adult (Fig. 1.21). Mature females lay eggs after copulation. Eggs hatch into wingless nymphs called hoppers. Hoppers moult five or six times and each time grow in size. The stage between moults is called as an instar. Wingless final instar (fifth or sixth) moults into winged adult (fledgling), and the process is called fledging. Adults do not show moulting and thus do not grow in size but increase in weight. Initially, adults that can fly are sexually immature but become sexually mature in about 10–13 days, copulate and lay eggs.

1.3.1 Egg Laying

The female lays eggs in bare sandy and moist soil. Female often determines the moisture in soil by probing the ovipositor before laying and, once it finds enough moisture, starts laying eggs in batches called egg pods (Fig. 1.22, A). The eggs appear like rice grains and are arranged as finger bananas (Fig. 1.22, B). Above the pod, which is about 3–4 cm long and 5–10 cm below the surface, a plug of froth is

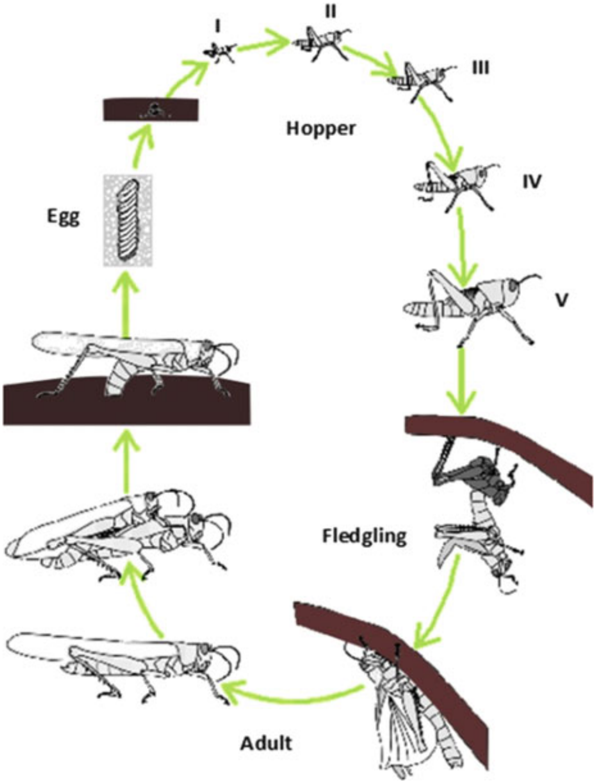


Fig. 1.21 Life cycle and different stages of desert locust. (Source: FAO 2001)

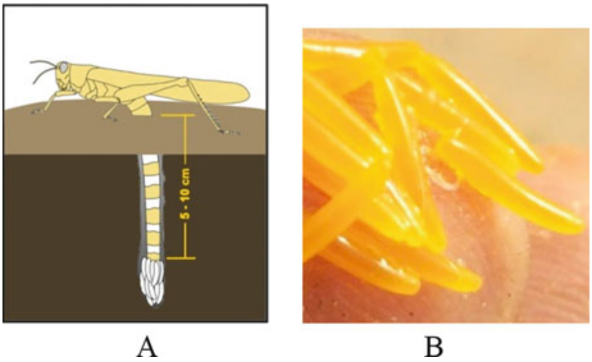


Fig. 1.22 (**a**) The female bores into the ground with the valves at the rear of her abdomen and lays a pod of eggs. The pod is about 3–4 cm long and is laid about 5–10 cm below the soil surface. (Source: FAO 2001). (**b**) Eggs like rice grains

laid. The gregarious phase locust lays pods with 80 eggs, whereas the solitarious phase lays pods with 90 to 160 eggs. Swarms lay egg pods with tens and even hundreds/thousands of pods per square metre. The scent and sight are important in attracting females to egg-laying females. Generally two to four egg pods are laid by females with the difference of ten days between the laying. However, due to natural mortality, a single female produces 16–20 viable locusts in a single generation.

The rate of development depends on the temperature of soil at pod depth and air. The mortality is more if the soil temperature is high. The survival of eggs up to hatching is also affected by habitat conditions, presence of egg parasites and predators. About 13 and 33% mortality is observed in solitarious and gregarious populations, respectively.

1.3.2 Nymphs/Hoppers

After incubation period is over, hatching occurs at dawn, and hatchlings make their way up through the froth plug to the surface. They immediately moult to the first instar. There are five instars in gregarious phase and sometimes six in the solitary phase. After fledging (or final moult), the young adult emerges. The rate of development of hopper is also dependent on temperature. However, the hoppers can manage their body temperature to a considerable extent by basking or seeking shade. There is no evidence that hoppers in relatively dry vegetation develop more slowly.

Hopper's Solitary Behaviour

Hoppers crawl over vegetation or ground and climb up to the top of plants before sunrise, and after sunrise, they descend from the plants to the ground. Hoppers climb back onto the vegetation during early morning and take shelter inside the plants or rest on plants. Hoppers descend again from the plants and bask on the sunny side of the plant in the afternoon. Near dusk, hoppers stop basking and climb back into the vegetation. The nature of habitat also influences the behaviour of hoppers. In bare land, hoppers most of the time move over the bare ground. In the area with uniform low vegetation, hoppers prefer moving along with movements under and out of the

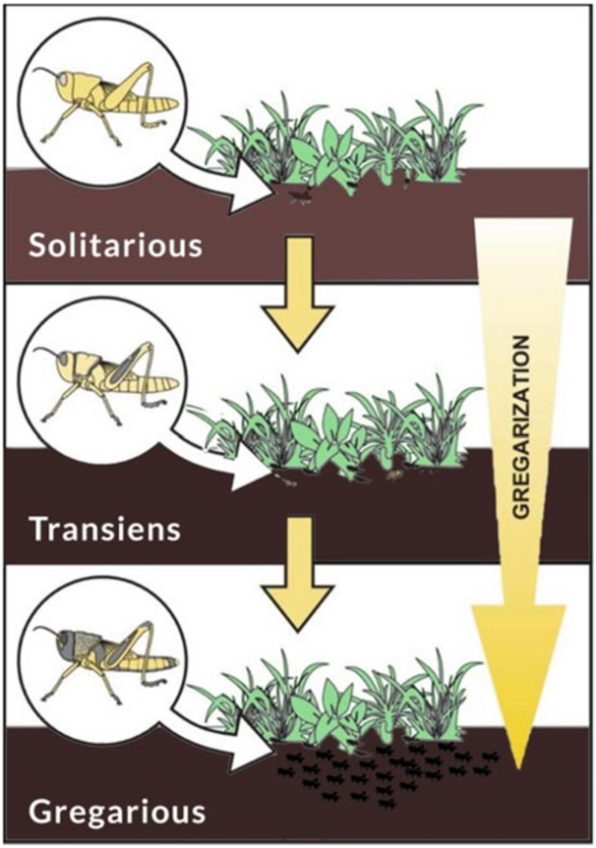


Fig. 1.23 When solitarious nymphs/hoppers of desert locust increase in number, their behaviour changes, and they become concentrated and can form groups. This often happens when vegetation starts to dry out. Their colour also changes and dark spots appear. (Source: FAO 2001)

vegetation, whereas in habitat with nonuniform vegetation, hoppers spend most of the time on vegetation instead of moving on the ground.

Hopper's Band Behaviour

When a number of hoppers increase in certain habitat, their behaviour is observed to be changed (Fig. 1.23). Hoppers are attracted during basking, feeding, roosting, moving on the ground or sheltering in vegetation and start forming groups in habitats that are less uniform with patches of dense vegetation and large bare soil areas. This grouping (transiens) can be regarded as an intermediate stage between hoppers of solitary phase and gregarious phase, whereas groups are less likely to form in habitats of uniform low vegetation and bare soil areas or uniform dense vegetation. Black markings on the green solitary hoppers also start appearing along with behavioural changes. Groups of hoppers found in the field are indication of onset of gregarisation and formation of bands. As the egg laying occurs at interval and all the eggs in a pod do not hatch at the same time, hopper bands are a mixture of instars in the same area at the same time of a recession period.

Shortly after hatching, hatchlings turn black and move to the nearest clump of vegetation but usually do not feed and move much for the first day and thus form small dense black patches of more than several thousand individuals per square metre (Fig. 1.24). Hoppers roost and march in an alternative manner during warm and sunny days. Densities of band depend on the behaviour stage of instars, habitat and weather. Hoppers on the ground form more dense bands as compared to roosting and marching hoppers. The density in bands on ground ranges from 30,000 to 1000 hoppers per square metre in case of first and fifth instar, respectively. However, the band area increases with age by merging of nearby bands together, and it continues till the fourth instar. But band area of fifth instar stage tends to be less cohesive. The movement of band is dependent on temperature, vegetation cover, size and consistency of the band about 200 to 1700 m in a day by bands of fourth instar hoppers. Usually, bands move during day, but night movements have also been reported on full moon night or exceptionally high temperature or in search of green vegetation. Heading path of band is often downwind and does not change, but at midday they are usually seen roosting under the vegetation.

1.3.3 Adults

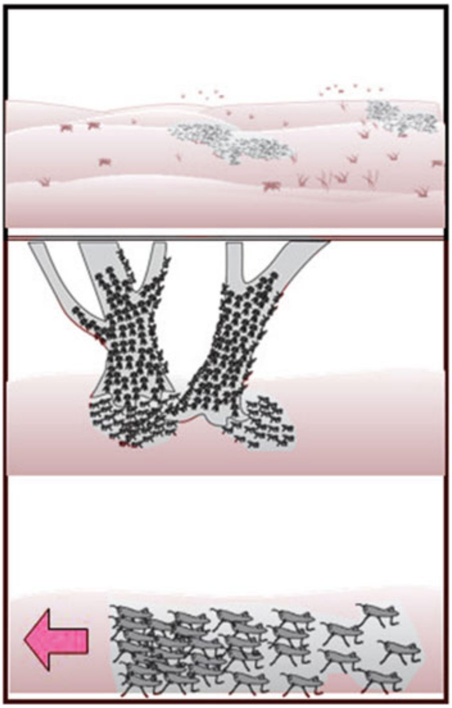
After fledging, wings of fledgling take about ten days to harden sufficiently to take sustained flight (Fig. 1.25). Initially, adults that can fly are sexually immature and remain immature until favourable maturation conditions, usually rainfall in already infested area or locust-invaded area. Maturation occurs in about 10–13 days under favourable conditions. It is well synchronised in swarms, as mature locust triggers immature locust to get mature. Mature adult females lay eggs within 3 weeks of fledging in a rainfed area with green vegetation and 35 °C temperature. Sexually Immature adults can survive for 6 months or more in dry conditions with ample of vegetation to eat, although in West Africa, south of Sahara, some are known to survive in winters, which are comparatively warmer with no breeding. Males mature sexually before females. The females remain sexually immature at the temperature below 20 °C.

Solitary Adults

A short-range displacement and migration due to brief low-level flight and sustained high-level flight, respectively, have been observed in solitary adult. The flights up to 1800 m have been detected by radar. The temperature range, 20–22 °C, for night flight is the same as that for day flight.

Gregarious Adults

Solitary adults also change their behaviour according to environment and numbers like the nymphs or hoppers do (Fig. 1.26). The increase in the number of solitary adults due to excessive previous breeding and shrinking of habitat due to dry conditions forces adults to concentrate in comparatively small areas suitable for their feeding and survival. This concentration of adults results in the formation of groups. Therefore, in addition to behavioural changes, change in the colour also



1. Shortly after hatching, hoppers form small dense black patches
2. These patches merge, forming larger bands
3. Hoppers in bands move together in the same direction

Fig. 1.24 Band behaviour of desert locust. (Source: FAO 2001)

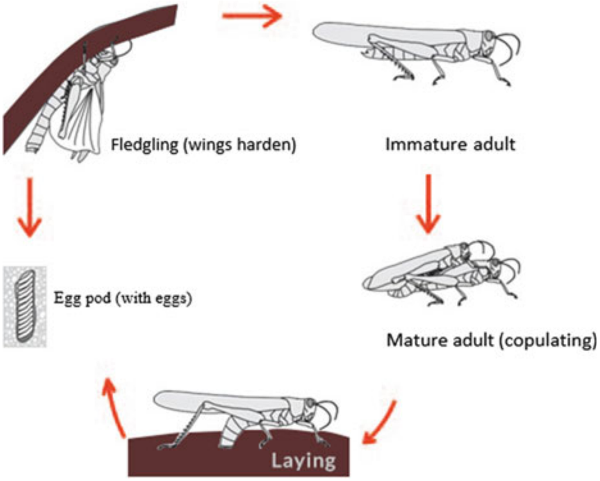


Fig. 1.25 On fledging, the adult's wings harden and the adult remains immature until maturation is stimulated by rainfall. In warm temperatures and good vegetation, it takes adults about 3 weeks to mature and lay eggs. If conditions are dry and cool, adults may remain immature for 6 months. (Source: FAO 2001)

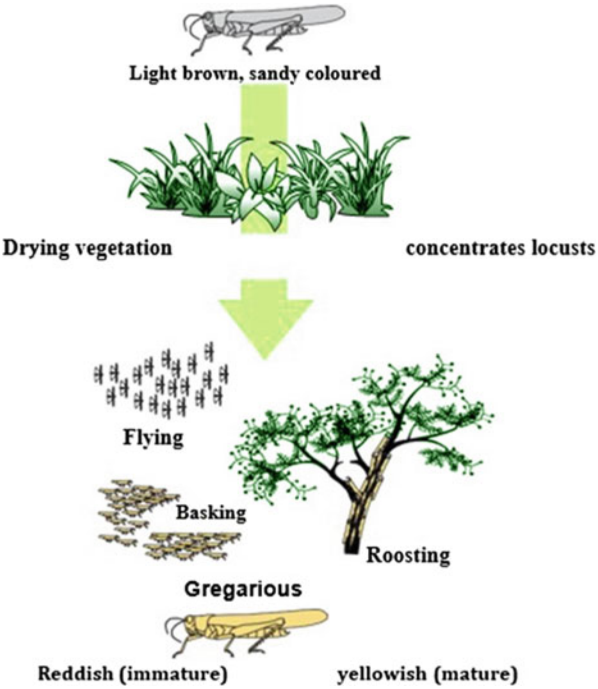


Fig. 1.26 When vegetation starts to dry up, solitary adults of desert locust may become concentrated and form groups. This can occur during basking, feeding, roosting and flying. During this process, the adult's behaviour and colour change. (Source FAO 2001)

occurs. Adults forming groups have traces of yellow unlike solitary adults with traces of pinkish colour on their abdomen and wings.

The adult grouping takes place during basking, feeding, roosting and even during flight where the wind and force tend solitary adults to concentrate. The presence of adult's groups in the field is an indication of being gregarious to make bands or swarms. Due to incoming adults, the swarm density is further increased.

1.3.4 Swarms

The first swarm is usually formed about tens or hundreds of kilometres downwind from the main breeding area. The adults are drifted away with the wind away from the breeding area; other nearby locusts around them also aggregate with them intensifying the swarm density. There are variable swarm densities. Generally, an average medium-density swarm may be comprised of about 50 million locusts per square km; however, the range may be 20–150 million per square km. During flight, the individuals forming the swarm spread out covering two to three times more area than that is covered during roosting. Swarms can occur in two forms: stratiform and cumuliform (Fig. 1.27). Stratiform swarms are flat and low-flying sheets of locust around tens of metres deep that occur during overcast cool weather or dawn. Cumuliform swarms, around 1500 m aboveground, are formed during warmer and

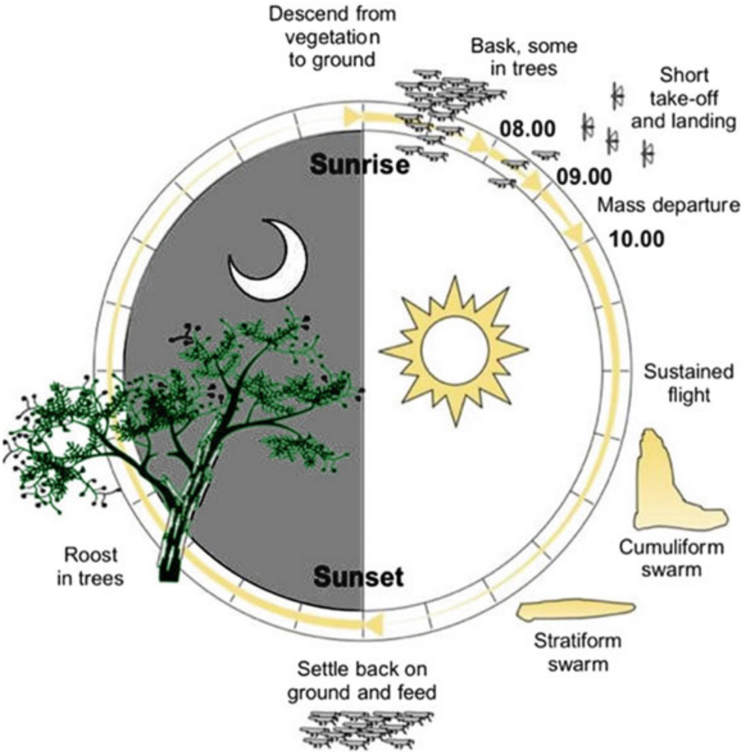


Fig. 1.27 Swarms of desert locust spend the night roosting in vegetation. At sunrise, they descend to the ground and warm up by basking in the sun. By mid-morning, swarms take off and will often continue flying until just before sunset when they land and feed. If the weather is unusually hot, swarms may settle at midday before flying off again in the afternoon. (Source: FAO 2001)

drier afternoon due to updrafts. Cumuliform swarms at higher levels form streams or swirling sheets.

In the locusts, glycogen in flight muscle could support flight for only 2–3 minutes. This may be adequate to support short flight from one plant to another, but for longer flights other sources of fuel are used. The stores of glycogen in tissue are meagre. However, haemolymph contains about 6 mg of trehalose that is the largest pool of available carbohydrate. In contrast, the quantity of lipid in the haemolymph is modest (2 mg), whereas in the fat body it may exceed 10 mg. Therefore, within 2 minutes of the onset of flight, adipokinetic hormone (AKH) is released by corpora cardiaca (CC), which stimulates the breakdown of fat body triglycerides into diglycerides supporting flights during swarms (Mordue et al. 1980).

Generally, swarm takeoff starts in the morning from the vegetation, where the swarms were roosting overnight (Fig. 1.27). They bask on the bare ground to get the greatest warming effect by exposing their bodies sideways to the sun. When the temperature increases, they take off but also land several times thereafter like an aircraft (Fig. 1.28). Landing of swarm is always by adults in the leading end, whereas takeoff is by adults in trailing end resulting in swarm in a rolling manner. For sustained flight during swarms, temperature is the key factor. The range of limiting temperature for sustained flight is 20–23°C.

The displacement of swarm is always downwind irrespective of the different directions of individual locust in a swarm. Swarms may fly up to nine or ten hours per day. The movement of swarm is slightly less than the wind speed and may cover about 100 km per day. In the absence of wind, speed of movement of swarm is very less. With the fall in temperature of the environment or an hour before the sunset, the swarm settles down and feeds whatever vegetation is available. However, young swarms sometimes continue flying after dark on warm evenings.

1.3.5 Survival

The female lays a pod of eggs into the soil with the ovipositor. The hoppers hatch and pass through five or six instar stages. At the final moult, they develop wings and become young adults. This phenomenon is known as fleding. Hopper development becomes faster at high temperature than at low temperatures. Sufficient vegetation develops due to rains with the egg laying and hatching of nymphs. However, only a few per cent of newly hatched nymphs survive to attain adulthood. Up to 70–80% of first instar nymphs die as a result of inadequate water reserve, cannibalism and predation. Further, 10–20% of hoppers die due to predation, cannibalism and other adverse conditions.

During recession period, the nymphs of all stages of solitary phase can be observed, because laying occurs at intervals and even in the same pod, and eggs do not hatch at the same time. Hence, hopper (nymphs) band is comprised of a mixture of instars.

1.3.6 Phase Polymorphism Theory

According to Sir Boris Uvarov (1921, 1966), phase polymorphism theory was enunciated stating that each species of locust can exist in two main forms (phases): the gregarious phase (phasis gregaria) and the solitary phase (phasis solitaria), which differ structurally, biologically and behaviourally. These are so distinct that earlier taxonomists regarded them as separate species. In between two phases, there occurs an intermediate phase or transitory phase (phasis transiens) during the transition of a population from one extreme to the other. If locusts are on the increase, they are referred to as congregans, and if they are on the decrease, they are called dissocians.

Solitary Phase The nymphs are variable in colour, green, grey or brown, or similar to the colour of its normal environment. The adults have longer and crested pronotum. The hind femur is comparatively longer than the fore femur. It has less prominent carina and rounded hind margin.

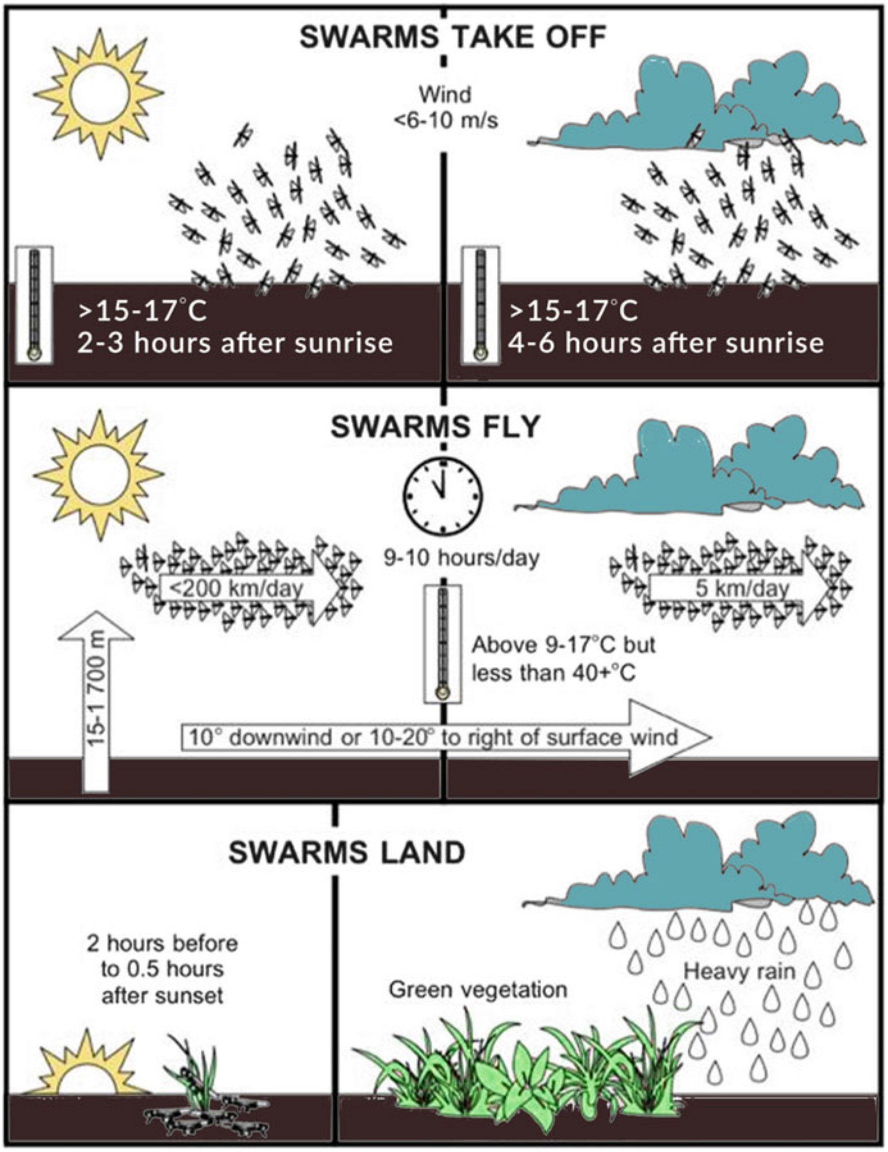


Fig. 1.28 Takeoff, flight and landing parameters of swarm desert locust. (Source: FAO 2001)

Gregarious Phase The nymphs have bold pattern of colouration, mainly black and yellow or orange. The adults have a shorter and saddle-shaped pronotum and relatively shorter hind femur. During the hotter part of the day, the nymphs or hoppers form large bands and march from place to place. The adults form large dense swarms, which fly over great distances under the influence of winds. The swarm settles with a fall in temperature (or sun may be hidden by clouds). Females are slightly longer (60–75 mm) than the males (55–70 mm) in length. Young adults are pale yellowish, and forewings have numerous brown-black spots; the hind tibiae are pale yellowish, except for the tip of spines. The median carina of pronotum at the curved hind margin is strong, and the subconical, prosternal median tubercle is present. Male possesses the genital lamina incised posteriorly. Various characteristics of solitary and gregarious phase are represented in a tabular form (Table 1.1).

During the recession period, the adults of the solitaria phase breed in the desert or 'outbreak' areas, and extensive and intensive breeding may lead to transformation of solitary phase to gregaria phase, leading to the formation of dense swarm. Biologically, the main difference between the two phases is the higher activity and gregarious tendency of the gregaria phase. Gregarious tendency manifestation in the nymphs is due to their habit of living in large bands, which march from place to place during the hotter part of the day. In adults, the gregaria forms occur in large and dense swarms, which may fly over great distances under the influence of winds. They continue to fly until environmental conditions (e.g. a fall in temperature) cause them to settle. The laboratory studies have revealed that nymphs reared in isolation (individually in separate container) are of the solitaria phase, while those reared in crowding together with many young nymphs lead to increased activity, which in turn is associated with the development of black pigment and other attributes of the gregaria phase.

Behavioural changes are rapid, like desert locusts reared in isolation in the laboratory; when first put into a cage with others, they try to avoid each other. During the process of avoiding, once they are touched with another locust, especially at the outer surface of hind femora, which results in attraction rather than repulsion and thus form groups and to switch from repulsion (the solitarious state) to attraction (the gregarious state), it takes only one hour. If crowded locusts become isolated, they revert to behave like in the solitaria phase, which is a slower process and may take several generations to complete the transition from gregarious to solitarious behaviour. Similarly, in the field also, several generations are required for solitary individuals to behave fully like gregaria during upsurges, when bands and swarms become progressively larger and closer. Hence, the gregarious and solitary (or solitarious) are used to indicate behaviour, gregaricolour and solitaricolour to indicate coloration, and gregariform and solitariform to indicate shape. However, morphological changes (changes in colour and shape) take more time. The change in colour from solitary to gregarious phase takes one generation, whereas the change in shape takes two or more generations. The change in rates of colour and shape associated with the change in phase may often be misleading as sometimes swarms of solitary locusts can be found in the fields. Ramesh Chandra et al. (1982) reported that insecticidal control applied to gregarious hoppers brought about a rapid phase transformation in desert locust. Reduction in the number of hoppers to a level below mutual stimulation due to mortality of most of the treated hoppers caused by the toxicants could be the only reason for the rapid shift to solitaria from gregaria within

Table 1.1 Comparative account of characteristics and morphometrics of solitary and gregarious phase

Characteristics	Solitary phase	Gregarious phase
Instars	5–6 (occasionally 4 or 7)	5
Colour	The nymphs (hoppers) are uniformly green in early instars but may be brown in last instars No black marking The adults are pale greyish brown; however, males change to pale yellow, when sexually mature. Females show no colour change	Initial instars of nymphs have black with pink markings, black with yellow or orange markings in late instars. Never green Adults are rosy pink on fledging and darken with age to greyish or brownish red and finally to yellow on sexual maturation
Eyestripes	6–7	6
Pronotum	Short, convex and crested	Longer, concave (saddle-shaped) and without crest
Antenna	27–30 jointed	26 jointed
Femoral spines	Both strong and weak	Only weak
Development period	30–39 days (summer breeding areas and Red Sea) 28–48 days (cooler periods, e.g. Northwest Africa)	25–57 days
Behaviour	No group or swarm formation in nymph Roost, bask, feed and move as individual adults fly as individuals	Nymphs form persistent and cohesive group bands Roost, bask and move together Adults are very mobile and fly as swarm by day hoppers move in bands
Morphometrics	E/F male 2.025 or less Female 2.075 or less F/C male 3.75 and more Female 3.85 and more	E/F male 2.225 or more Female 2.272 or more F/C male 3.15 and less Female same like male

Note: *C* width of head, *E* length of forewing, *F* length of hind femur
Measuring unit mm
Source: Srivastava (1996) and Usmani and Usmani (2018)

a short time. Desert locust hoppers with stabilised gregarious characters changed to transient solitaria before they fledged (Fig. 1.29).

The view that the more dispersed individuals lead to the solitary phase and dense population leads to gregarious phase, which is a reversible process, is declined today. Recent researches have revealed that corpus allatum hormone or juvenile hormone (JH) is more in solitary individuals than in the gregarious forms throughout life. Further, the prothoracic glands do not disappear in the adult; JH and the neurosecretory cells (NSC) in the brain act on the reproductive system at the same time. Therefore, the gregarious forms are more mature in terms of structure, physiology and behaviour than the solitarious forms. Similarly, visual, mechanical and chemical stimuli (diffusion of specific substances), availability of suitable food, oviposition sites, etc., also influence the process of gregarisation. Further, the black gregaria nymphs that absorb more radiant heat have high internal temperature, leading to a high level of activity in the gregaria phase. The suitable natural

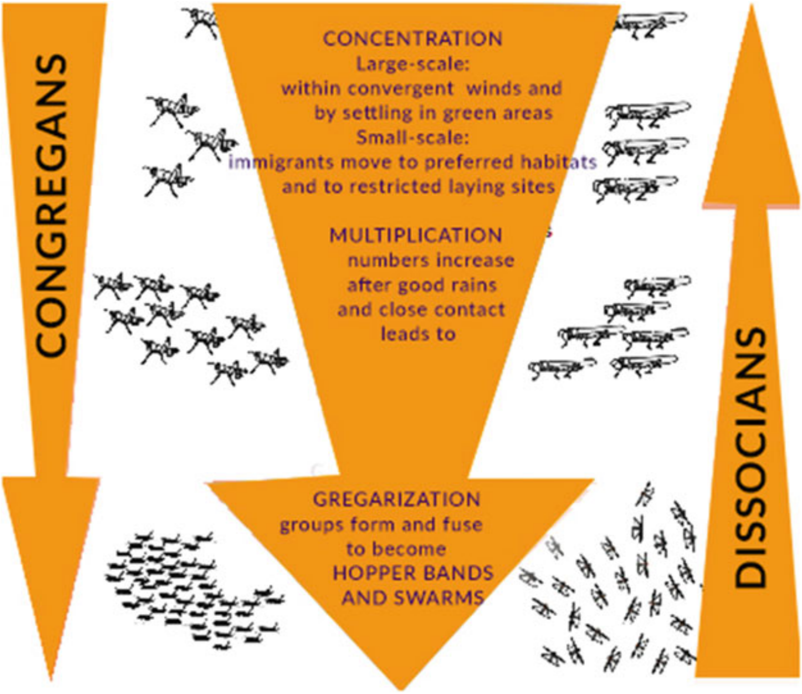


Fig. 1.29 Desert locusts have the ability to change their behaviour, physiology, colour and shape in response to a change in locust numbers. At low numbers, locusts behave as individuals (solitarious phase); at high numbers, they behave as a single mass (gregarious phase). Precise thresholds at which these changes occur are not established. Three processes are involved in phase transformation: concentration, multiplication and gregarisation. (Source: FAO 2001)

conditions induce crowding and therefore cause gregarisation and lead to locust plagues in restricted regions (outbreak areas), where as a result of flooding or variable rainfall, the habitats favourable to breeding are liable to irregularly periodic contractions in size. Therefore, swarms may give rise to solitaria or gregaria forms according to the local conditions of invasion area where they breed.

Thus, it is evident from the theory of phase polymorphism that locusts are the grasshoppers that are changed morphologically and behaviourally under favourable environmental conditions on crowding (Fig. 1.30). Thus, all locusts are grasshoppers, but all grasshoppers are not locusts. From a taxonomic viewpoint, the species of locust and grasshopper are not different; the basic difference is that whether a particular species of grasshopper is capable of forming a swarm during crowding under suitable environmental conditions or not. The solitary and gregarious phases are also regarded as solitary and nomadic morphs. Overcrowding leads to swarming behaviour. During a hot day, the absorption of radiant energy causes the elevation of body temperature; as a result, physiological stimulation of the hindlegs causes an increase in the secretion of serotonin (Morgan 2009), leading to the change

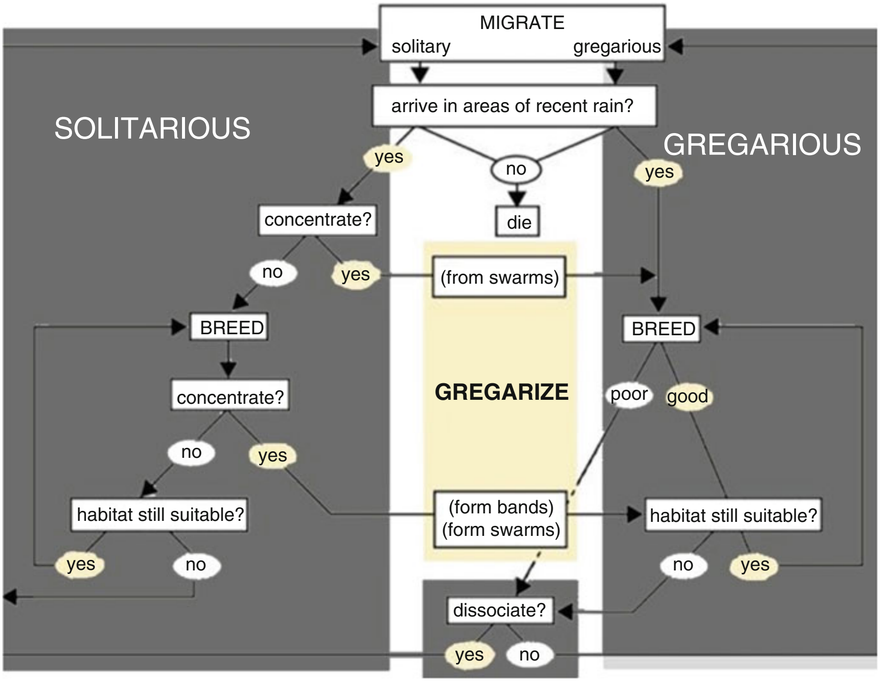


Fig. 1.30 Summary of desert locust population dynamics, showing the influence of the environment on locust behaviour and phase. (Source: FAO 2001)

in body colour of the locust. Thus, due to increased metabolic rate, they eat more and breed more. The nymphs of solitary phase do not pose a major economic threat to agriculture or forest ecosystems. During favourable conditions, more secretions of serotonin are released, which leads to a dramatic set of changes; as a result, more breeding takes place. When increased number of hoppers, move in bands, lead to gregarious phase on maturation into winged adults.

1.3.7 Migration and Seasonal Distributions

The change in wind flow due to seasonal changes causes migration of locusts in a particular area at particular zones as displacement of both day-flying swarms and night-flying solitary individuals is downwind. Therefore, at the beginning of summer, a southward movement has been observed from Northwest Africa into Sahel of West Africa. Similarly, a northward movement has been observed during autumn although movements of night-flying solitary locusts are restricted due to low temperature as compared to day-flying swarms. As the seasonal wind pattern changes the environment of breeding habitat, the migration of locusts is rapid and up to a great distance.

Likewise, the rain is also one of the factors for the downwind displacement to rainfed area, for example, in summer, Sahel of West Africa and Sudan and, in winter, the Red Sea coasts are preferred. Moreover, on the basis of winds and temperature, desert locust swarms move in the southern circuit, i.e. from the Sahelian belt of West Africa to southward movement.

1.3.8 Recession

When the locust density is low in semiarid or arid areas away from main agriculture zones and the hopper bands and swarms are rare or absent, there is no significant crop damage. This period is known as recession, and the area in which these populations are confined is known as the recession area. The recession area is an appropriate one for the surveillance of locust breeding and early control of locust population. As the swarms fly out of the recession area, the locust breeding may increase if heavy and appropriate rains occur in that area. Recession area is estimated to be about 16 million km^2, which includes around 30 countries.

1.3.9 Outbreak

The transition from recession situation to a plague is called the outbreak. An outbreak occurs when there is tremendous increase in locust numbers by multiplication, gregarisation or congregation of locusts over several months and bands or swarms form if not controlled. The population of hoppers during early stages of an outbreak is widely distributed and forms low-density swarms. According to Uvarov (1951), the ultimate aim in locust control should be alteration in ecology of outbreak area so that the process of gregarisation may be hampered. However, this concept has changed recently to study the behaviour to prevent swarming.

1.3.10 Upsurges

An upsurge occurs when successful breeding of a large number of generations are produced by an initially small population with each generation, and a total number of hoppers, hopper bands and swarms increase. Several outbreaks that occur at the same time may lead to an upsurge. During upsurge, there is a migration from breeding area to the other, and usually most of the upsurges die before turning into plagues. The occurrence of several upsurges at the same time in the same area has also been observed.

Upsurge prevention was developed in association with persisted pesticides (organochlorines) and is based on the hypothesis that any outbreak can lead to an upsurge. It is advocated that destruction of the initial gregarious population stops the development of upsurge and thus prevents a plague formation.

1.3.11 Plagues

When there is widespread and heavy locust infestation either in the form of bands or swarms, it is called a locust plague (Showler 2013). A major plague exists when two or more regions are affected simultaneously. The locust plague occurs when breeding conditions are favourable and control operations are unable to prevent local outbreaks from developing into upsurge. Plagues are separated by recession periods. In the 1900s, six major plagues were reported, and one out of them lasted for almost 13 years. Plagues cover about 29 million km^2 area and about 57 countries. After a period of expansion and increasing swarm size, the plagues decline taking up to 2 years to end; decline may also depend on the region. The ninth plague ended in 1989, followed by a major upsurge in 1992–1994. The main factors which play important role in ending plagues are:

(i). Insufficient rainfall
(ii). Movement of swarms to areas unsuitable for breeding and survival of the locusts
(iii). Control efforts, predominantly based on chemical methods

Plague suppression occurs when an upsurge becomes widespread and the swarms become vast and numerous. Efforts are concentrated to have greatest impact on locust populations. This strategy also involves crop protection activity by farmers in cultivated crops (Symmons 1992).

1.3.11.1 Declines

Decline in plagues occurs due to natural factors, like failure of rains, control operations by humans or migration of adults or progeny. The shortage of rains in the Horn of Africa in 1955 caused a decline in plague since then. Similarly, a decline due to swarm migration from transatlantic was observed in October and November 1988. Human intervention also plays an important role in bringing locust plagues to an end through various control operations.

1.3.11.2 Permanent Habitat

The desert locust habitat is comprised of African countries, north of the equator, the Mediterranean Europe, Iran, Kazakhstan, Azerbaijan, Turkmenistan, Uzbekistan, Tajikistan, Afghanistan, Siberia and the soil of rivers ending in Caspian Sea, Lake Balkhash, grasslands, etc. According to FAO, the breeding of desert locusts occurs in deserted areas of South Arabia, Kuwait, Yemen, Iran, Oman, Kenya, Ethiopia, Somalia, Pakistan and Egypt. The developing swarms enter in India generally through Pakistan.

1.3.11.3 Nature and Extent of Damage

The effect of locust attack on growth and yield are complex and variable due to pest feeding behaviour, its density and distribution within the crop and timing of attack in relation to plant growth. Seedling and milky grain stages are the most vulnerable stages for cereals (Wright 1986). The feeding behaviour and damage caused by locusts is not uniform or predictable (Uvarov 1977). There may be variations in the loss caused in between or within the individual field (Jago 1993). The activity of locust swarm is mainly regulated by temperature, being most active at an optimal body temperature between 35 °C and 40 °C. At low temperature, the activity is declined and ceased below 15 °C; even then, they can damage the crops or vegetation. The desert locust does not spare a single crop or vegetation, leading to a serious impact on livestock, because of unavailability of enough barley, citrus crops, date palm, millet, sorghum, vegetables and fruits, which are needed for food by human beings and their cattle. About 30 million square kilometre area is invaded by it, and in about 50% of this area, breeding occurs. The invasion belt of this locust ranges from north and eastern Africa to Assam in India. In between this range, about 57 countries are represented, having 20% of the world land area.

In India, the locust swarms reach Assam in east and Kerala in the south. Migrations are seasonal. Due to rain in different areas at different times, the breeding takes place during summer or during spring season. During monsoon rains, breeding takes place in northwestern part (Thar Desert) of India and adjoining parts of Pakistan, and the resultant swarms from August onwards migrate as far as west Iran and Arabia. With the onset of rains during winter-spring in this region, breeding takes place leading to swarm formation, which migrates to Indo-Pakistan region during May to July. In India, the recent swarms have been continued since November 2019.

1.4 Plague Cycles in India

The locust plagues occur in a sort of cycle, in which plague period is followed by recession period. Swarm production and damage that takes place due to widespread breeding during consecutive years is referred to as plague period. It may be followed by a period during which there is a little or no activity of hoppers, and this is known as recession period. In India, in general, and in Rajasthan, in particular, there have been many desert locust plagues, starting from 1964 (Table 1.2). The latest desert locust plague which invaded India started on 21 May 2019 and is still continuing in various parts of northwest and central India. Very recently, there are reports that locust swarm reached Sawai Madhopur, Kota, and Nagore, besides Barmer, Jaisalmer, Jodhpur and Bikaner regions; it also reached Jaipur (Fig. 1.31). Swarms of desert locust including immature, maturing and mature adults were observed at various districts of Rajasthan, Gujarat, Madhya Pradesh and Uttar Pradesh during from 1st June to 15th June, and control operations were undertaken at 241 spots out of 659 number of spots, covering 33,867 hectare area (Desert locust situation bulletin 2020). Desert locust attack occurs as a ‘cycle’—a period of 5–6 years called the plague period in which due to widespread breeding swarm production and damage to crops occur, and this is followed by a period of 1–8 years with little activity called as recession period. Recession period is again followed by another spell of plague and so on. In India, ten such locust plagues at intervals of 1–8 years have occurred since 1963 (Sharma 2014).

1.5 Various Locust Organisations

The following organisations are engaged in providing all information about the hoppers and adults of locusts.

1.5.1 Locust Warning Organization (LWO)

This organisation comes under the Ministry of Agriculture, Government of India. It works under the direct control of Plant Protection Advisor to the Government of India, Directorate of Plant Protection, Quarantine and Storage, Ministry of Food and Agriculture, New Delhi. Various programmes about research and various locust control strategies are implemented by it. Its state head office is located at Jodhpur. There are circle offices located at Barmer, Bikaner, Suratgarh, Palampur, Bhuj, Jalore, Nagaur and Churu. Its central headquarter is at Faridabad.

The main objective of this organisation is to conduct survey and monitor the locust (hopper and adult) activity and issue warning to concerned persons (Anonymous 2019). Locust control operations and strategies are also important tasks of this organisation. This organisation also conducts programmes to train farmers and village workers. To conduct field-oriented research and biology of locusts, it is useful in integrated pest management strategies of this serious pest.

1.5.2 Field Station for Investigation on Locust (FSIL)

The Ministry of Agriculture, Government of India, established FSIL at Bikaner under Directorate of Plant Protection, Quarantine and Storage in 1957. The main purpose or objective for establishing this centre was to conduct research under field conditions and simulated laboratory conditions on various aspects of locusts and grasshoppers, such as biology, ecology and various control measures for locusts.

1.5.3 International Cooperation Programmes (ICP)

The main objectives of this programme are:

- To exchange information on status of locust hoppers and adults between various countries where the outbreak areas of locusts occur, such as Pakistan and other adjoining countries.
- To organise and participate in meetings conducted by FAO’s desert locust control committee.

Table 1.2 Year-wise locust upsurge data of India from 1963 to 2020

S. No.	Year	Locust infestation	Action taken	Remarks
1	1963	No infestation	Nil	Nil
2	1964	Upsurge	Control operations organised	Successfully controlled
3	1965	No infestation	Nil	Nil
4	1966	No infestation	Nil	Nil
5	1967	No infestation	Nil	Nil
6	1968	Upsurge	Control operations organised	Successfully controlled
7	1969	No infestation	Nil	Nil
8	1970	Upsurge	Control operations organised	Successfully controlled
9	1971	No infestation	Nil	Nil
10	1972	No infestation	Nil	Nil
11	1973	Upsurge	Control operations organised	Successfully controlled
12	1974	Upsurge	Control operations organised	Successfully controlled
13	1975	Upsurge	Control operations organised	Successfully controlled
14	1976	Upsurge	Control operations organised	Successfully controlled
15	1977	No infestation	Nil	Nil
16	1978	Upsurge	Control operations organised	Successfully controlled
17	1979	No infestation	Nil	Nil
18	1980	No infestation	Nil	Nil
19	1981	No infestation	Nil	Nil
20	1982	No infestation	Nil	Nil
21	1983	Upsurge	Control operations organised	Successfully controlled
22	1984	No infestation	Nil	Nil
23	1985	No infestation	Nil	Nil
24	1986	Upsurge	Control operations organised	Successfully controlled
25	1987	No infestation	Nil	Nil
26	1988	Upsurge	Control operations organised	Successfully controlled
27	1989	Upsurge	Control operations organised	Successfully controlled
28	1990	Upsurge	Control operations organised	Successfully controlled
29	1991	No infestation	Nil	Nil
30	1992	No infestation	Nil	Nil
31	1993	Upsurge	Control operations organised	Successfully controlled
32	1994	No infestation	Nil	Nil
33	1995	No infestation	Nil	Nil
34	1996	No infestation	Nil	Nil
35	1997	Upsurge	Control operations organised	Successfully controlled
36	1998	No infestation	Nil	Nil
37	1999	No infestation	Nil	Nil
38	2000	No infestation	Nil	Nil
39	2001	No infestation	Nil	Nil
40	2002	No infestation	Nil	Nil
41	2003	No infestation	Nil	Nil
42	2004	No infestation	Nil	Nil
43	2005	Upsurge	Control operations organised	Successfully controlled

(continued)

Table 1.2 (continued)

S. No.	Year	Locust infestation	Action taken	Remarks
44	2006	No infestation	Nil	Nil
45	2007	Upsurge	Control operations organised	Successfully controlled
46	2008	No infestation	Nil	Nil
47	2009	No infestation	Nil	Nil
48	2010	Upsurge	Control operations organised	Successfully controlled
49	2011	No infestation	Nil	Nil
50	2012	No infestation	Nil	Nil
51	2013	No infestation	Nil	Nil
52	2014	No infestation	Nil	Nil
53	2015	No infestation	Nil	Nil
54	2016	No infestation	Nil	Nil
54	2017	No infestation	Nil	Nil
56	2018	No infestation	Nil	Nil
57	2019	Upsurge	Control operations organised	Successfully controlled
58	2020	Upsurge	Control operations organised	Efforts on to control

Source: FAO

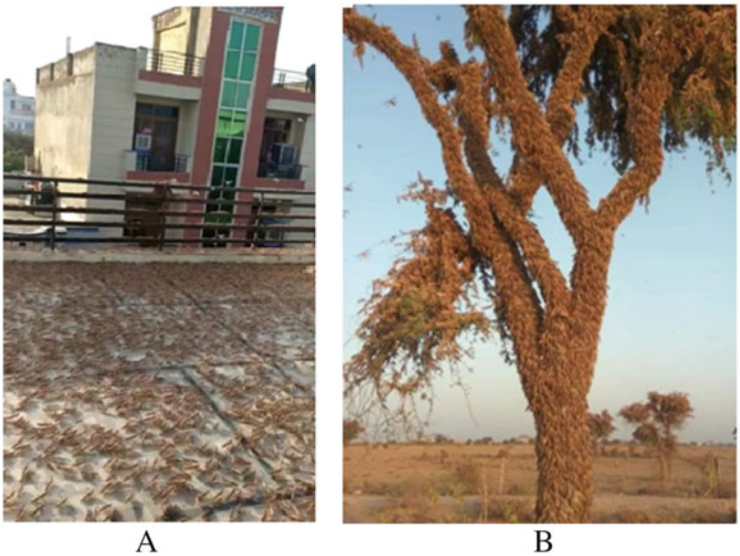


Fig. 1.31 (**a**) Desert locust swarm invaded in residential area at Jaipur, Rajasthan, India. (**b**) Tree with locust swarm

- Formerly a fortnightly bulletin was published by the Ministry of Agriculture, Government of India. Now this task is performed by wireless and e-mail services.

- The warnings are conveyed to make necessary arrangements for the management of locust problem.
- For successful control and management of locusts, international cooperation is essential. From this point of view, there is a convention between the Government of Pakistan, Iran and other adjoining countries with the Government of India. The main aim of this cooperation is to exchange information regarding the locust situation (the locust warning organisation established by the Government of India keeps a careful watch on locust status); to collect information from various states and countries; and to convey subsequent messages to all concerned through periodical bulletin, radio broadcasts and e-mails.

1.6 Locust Control and Management

The first international locust conference was held in Rome in 1920, and during a major plague in 1925–1934, many affected countries agreed to cooperate. In 1930, the Anti-locust Research Centre (ALRC) was established in London. The International Locust Information Network, which was also established in 1930, is now maintained by FAO. In 1940, the UK established an Interdepartmental Committee on Locust Control in London, which coordinated and financed locust control. After the World War II, the governments of Egypt, India, Iran and Pakistan continued to maintain permanent anti-locust organisations for mutual cooperation.

Since locusts are very devastating, their control is quite essential. As locusts may cause almost 100% loss of the harvest by invading the crop fields, it becomes a matter of life and death for the farmers. The locust control is carried out at every stage of its development.

Eggs The locust eggs can be destroyed by many mechanical methods, like ploughing, harrowing and hand digging the egg pods or by flooding the fields.

Nymphs The nymphs are the most vulnerable stage for destruction. Various methods can be employed for destroying nymphs:

(i) Digging trenches of 2 × 2 feet around the breeding ground, when the nymphs hatch out, they can be derived into the trenches and can be burnt or buried into the soil. Some pesticides may also be used to kill the emerging nymphs.
(ii) The hoppers can be burnt with flame throwers, when they are resting on the bushes during dark.
(iii) The use of poison baits can destroy the nymphs/hoppers. The poison baits were prepared by sodium arsenite, sodium fluosilicate, Paris green or aldrin or any other insecticides + bran or saw dust + enough water. The hoppers feeding the baits were destroyed.
(iv) Pesticides—formerly 5% BHC dust or 0.2% aldrin—were used to kill the hoppers, but now newer pesticides, like chlorpyriphos 20% EC or synthetic pyrethroids, may be used.

Fig. 1.32 Vehicle-mounted Micronair. (Source: Micron Group)

Adults Various methods employed to control adults are:

(i) The adults can be swept and buried in the soil, or they can be killed by beating with thorny sticks.
(ii) The adults can be burnt using flame throwers, particularly during night or early morning, when they are sluggish due to comparatively low temperature of the environment. During cold weather, the adults generally rest on the top of the trees during night. They fall onto the ground by beating the twigs and can be either destroyed by beating or burnt or buried.
(iii) During daytime, the swarms can be prevented by beating drums creating loud sound or by waving white pieces of cloth, which prevents the swarms from settling on the crops.
(iv) During flight, the swarming locusts can be burnt by flame throwers. Insecticidal sprays can be used by vehicle-mounted Micronair (Fig. 1.32) and Ulvamast (Fig. 1.33), handheld spinning disc sprayers (Fig. 1.34); examples of such sprayers are Micron Sprayers, Micro-Ulva and Ulva+, Berthoud C5 and Goizper and an aircraft. Chlorpyriphos 20% EC or some synthetic pyrethroids can be used to kill the locusts on the ground surface.

In addition to the activities of the Locust Warning Organization (LWO) and other NGOs, such as Gramodaya Mission, Mahan Bharat Abhiyan and Saral Jeevan India Foundation, Hyderabad, and the Food and Agriculture Organization of United Nations (FAO), certain guidelines are prescribed keeping in view the current scenario of locust swarms for delegates on application techniques and safe and judicious use of pesticides for locust control:

(i) The objective should be destruction of locust eggs through mechanical means, like ploughing, harrowing and trenching, or by using quality pesticides as approved by the Central Insecticide Board and Registration Committee (CIB

Fig. 1.33 Vehicle-mounted Ulvamast. (Source: Micron Group)

Fig. 1.34 Battery-operated Ulva hand sprayer or handheld sprayers

& RC) (Table 1.3). Presently, the recommended pesticides are chlorpyriphos EC (20%) and lambda-cyhalothrin EC (5%) for use in cropped area. Local measures may include the application of the neem seed kernel (NSK) extract, etc.

(ii) Efforts on capacity building on locust diagnosis and management for “farmer bodies” are imparted by concerned state and central agencies. They will supplement the efforts of locust control agencies for effective containment.

(iii) Based on the local/national news on swarm movement, localised survey for identifying locust eggs and nymphs may be undertaken. E-app could also be used for monitoring and forecasting of swarms.

(iv) Inform concerned local/national authorities on swarm movement to enable them to guide the community and initiate control action based on the infestation.

(v) For destruction of locust eggs, besides using mechanical measures, digging trenches of 2 × 2 feet will make the nymphs fall down in trenches, which could be buried, preventing their further expansion.

Table 1.3 Central Insecticide Board and Registration Committee (CIB & RC)-recommended pesticides for the control of desert locust

S. No.	Chemical	Dose (Gram active ingredient per ha.)		Dose of the formulation/ha
		Hoppers	Adults	
1	Bendiocarb 80% WP	100	100	125 gms
2	Chlorpyriphos 20% EC	240	240	1.2 litres
3	Chlorpyriphos 50% EC	240	240	480 ml
4	Deltamethrin 2.8% EC	12.5	12.5	450 ml
5	Deltamethrin 1.25% ULV	12.5	12.5	200 ml
6	Diflubenzuron 25% WP	60	NA	240 gms
7	Fipronil 5% SC	6.25	6.25	125 ml
8	Fipronil 2.92% EC	6.25	6.25	216 ml
9	Lambda-cyhalothrin 5% EC	20	20	400 ml
10	Lambda-cyhalothrin 10% WP	20	20	200 gms
11	Malathion 50% EC	925	925	1.850 litres
12	Malathion 25% WP	925	925	3.7 kgs

(vi) The quality pesticides (dusts and liquid) as approved by the Central Insecticide Board and Registration Committee (CIB&RB) are used.
(vii) Local measures with neem seed kernel, acoustics (drum beats) (Fig. 1.35), making vegetable matter inedible.
(viii) Forming teams to delimit the infested area spray downwards and not mixing water in ULV-formulated pesticides.
(ix) Sensitise other farmers to take preventive measures to avoid pesticide accidental toxicity.
(x) Utilise drones provided by approved agencies for chemical control, taking all precautionary measures.

Further, many other suggestions regarding locust control may be discussed.

Most adverse side effects of spraying can be avoided by adhering to the FAO application guidelines and careful choice of insecticides to be sprayed. Regular training course for technicians, operators and other locust control personnel in survey, pesticide safety, storage and handling of pesticides is essential.

Preventive control, i.e. checking the buildup of locust population as a whole to avoid risks to agriculturists, has been the policy of FAO for approximately 30 years. However, this policy was not successful in two major upsurges, i.e. 1985–1989 and 1992–1994. The areas involved are enormous, when breeding conditions are favourable. It may be impossible to control a major proportion of the locust population to prevent a developing upsurge. The problem of targeting control measures is both technical, lack of decision linked to socioeconomic and political impact data and cost of different options. Currently, there is little consensus amongst technical expert donors and affected countries on how to proceed. A number of donors support

Fig. 1.35 Drum beating

long-term research and development projects in addition to emergency control operations. The US Agency for International Development (USAID) has brought these functions together under the Africa Emergency Locust and Grasshopper Project.

It is essential that the countries affected and donors find means to unite behind an agreed long-term management strategy for desert locust control. This strategy should be based on the principle of integrated pest management (IPM), so that the risks associated with desert locust are assessed and mitigated by a variety of available measures. These initiations are needed at national, regional and international levels, and these need to cover:

(i) Integrated information and early warning system
(ii) Contingency plan in the event of threatened upsurge
(iii) Cross-border agreement

Timely reporting by affected countries regarding locust habitat and distribution and control operations is very important for further guidance by regional and international efforts.

Regional cooperation for anti-locust efforts is very essential, including sustainable cost-sharing arrangements by the locust-affected countries. International intervention for desert locust control is also needed to ensure:

(i) Required flexibility to resource campaign
(ii) Consistent and cost-effective control
(iii) Targeted control on the basis of social, economic and environmental considerations

When internationally coordinated campaign becomes necessary, it should be rapid and effective. Donors should establish the resource and systems for effective campaign well in advance including activation of consolidated regional funds and ensure rapid access to appropriate chemical control agents.

Desert locust breeding and seasonal migration pattern cannot be prevented by any national boundary; hence, it is only with international cooperation that long lasting solutions can be found for prevention, control and suppression of locust plagues. Crops and grazing areas in about 20% of the Earth's surface, including 65 countries in Africa, Middle East and southwestern Asia, are subjected to swarm invasion (Fig. 1.1).

Initial outbreak areas are in remote semiarid areas of Sahara, Arabian Peninsula and along the border of India and Pakistan. Plagues begin when exceptional rainfall occurs in potential outbreak areas simultaneously; under these circumstances, a generalised upsurge in the desert locust population may take place. Consecutive seasonal breeding rarely occurs in the same sites in successive years or even successive decades (FAO 1968).

In between plagues, the desert locust number and density is low and they are found only in drier central part of total area. This smaller zone is known as the recession area. It is comprised of more than 25 countries extending from Mauritania to northwest India.

Technical Challenges to Strategies

Modern locust studies and control strategies started in 1920. According to the findings of Uvarov (1921) and Faure (1923), solitary and gregarious locusts were one species, not two. The technical challenges needed are:

(i) To acquire sufficient knowledge on biology and behaviour of desert locust
(ii) To find its breeding habitat
(iii) To forecast locust population density and migration
(iv) To develop methods to stop potential plagues

Locust control, like all other pest management, aims at protecting agricultural production. In 1930 and early 1940, the locust control involved individual farmers protecting their crops by using physical means, such as burning, beating, building barriers or digging trenches, but later besides the sole aim to protect crops, the strategy began to attack the locust, wherever possible, in order to prevent the swarms and plagues. Current information on locust breeding, density and phase is the starting point of all locust control operations. The basic needs are:

(i) Continuous ground and aerial survey of potential locust breeding areas
(ii) Concurrent data on rainfall and vegetation
(iii) Collection, analysis and exchange data across affected countries
(iv) Capacity for rapid assessment of the need for locust control based on the above

Chemical Control and Pesticide Choice

(i) Once the locust breeding is detected, locust control operations consist of a range of chemical control techniques applied on the basis of availability of chemicals, equipment and persons deputed at remote location and over a relatively short span. After the publication of the *Silent Spring* by Rachel Carson in 1962, there was a complete ban on synthetic organic pesticides in the USA and other developed countries. DDT and BHC dusts were used for aerial use by planes in breeding areas to damage eggs and newly hatched nymphs. DDT and BHC were banned in 1993 everywhere. The locusts often reach farmer fields, and in the absence of any other technology, farmers have no choice but try to save their crops by traditional methods, such as building barriers, digging trenches, burning, beating and smoking. Conventional insecticides used in locust control (Table 1.4) are generally neurotoxins with active ingredients divided into four groups of organochlorines—DDT, BHC, dieldrin, endrin—which were broad spectrum and persistent in the environment. Previously, the category of choice was now banned by many countries. These toxicants were very persistent and remain for a long period, thus contaminating the environment, and were cost-effective also.

(ii) Organophosphates—it includes two most widely used insecticides: malathion, fenitrothion, etc. They are moderately fast in action and relatively nonpersistent but nonselective compounds. Malathion possesses a very low mammalian toxicity.
(iii) Carbamates—examples are bendiocarb: similar in action with organophosphates. Like organophosphate compounds, their mode of action is by acetylcholine accumulation at synaptic clefts.
(iv) Synthetic pyrethroids—examples are deltamethrin, fenvalerate and lambda-cyhalothrin, which are fast acting (knockdown within minutes), varying level of persistence and broad spectrum. They are of fairly low mammalian toxicity.

Difficulties experienced in chemical control campaigns are poorly maintained application equipment, wrong application techniques and adverse meteorological condition at the time of spraying.

The formulations of insecticides include emulsifiable concentrate (EC), ultralow volume (ULV) spray of oil-based pesticides, baits and dusts that are most likely to be used by the farmers.

ULV spraying can be used in one or more ways depending on the life stage of locust:

(i) Band spraying (for individual hopper)
(ii) Block spraying
(iii) Settled swarm spraying
(iv) Flying swarm spraying: The locust may get entry into the fan of planes and lead to accident. It is potentially the more rapid and pesticide efficient spray strategies (Symmons 1993).

Table 1.4 Locust control campaign in India

Year	Period	Type of campaign	Area treated (ha)	Pesticides used (kgs/lit)	Pesticide
1993	July to October 1993	Yellow/pink desert locust hoppers and swarms (190 Nos) (Jaisalmer, Barmer, Jalore, Bhuj)	310,482	688,255 30,934 47,577 36,860	BHC 10% dust Dieldrin 18% Malathion ULV Fenitrothion ULV
1997	July to October 1997	Yellow/pink desert locust hoppers and swarms (04 Nos) (Jaisalmer & Barmer)	23,596	7974 3660	Fenitrothion ULV Malathion ULV
2002	July 2002	Migratory locust population (Jodhpur)	42	42	Malathion 96% ULV
2005	September to December 2005	Loose pink swarm and hoppers (Jodhpur, Bikaner, Jaisalmer)	16,640	10,476 1883	Malathion 96% ULV Fenitrothion 96% ULV
2007	April to September 2007	Loose pink/yellow swarm and hoppers (Jodhpur, Bikaner, Jaisalmer)	536	536	Malathion 96% ULV
2010	October to November 2010	Hoppers/fledgling (Jaisalmer)	4700	4700	Malathion 96% ULV
2016	June 2016	Migratory locust, etc. (Leh area of J&K)	1205	1928	Chloropyriphos 20% EC
2016	November 2016	Tree locust, etc. (Jodhpur)	40	40	Malathion 96% ULV
2017	November 2017	Tree locust, etc. (Jodhpur)	40	40	Malathion 96% ULV
2019	December 2019	Desert locust	310,584	276,505	Malathion 96% ULV
2020	June 2020	Desert locust	89,905	–	Malathion 96% ULV

Source: LWO Jodhpur 2019

Pesticide Application

Until 1980, spraying with barriers of persistent organochlorine insecticides (mainly dieldrin) for control of hoppers was used. The insects were often killed weeks or months after spraying, by eating vegetation in such barriers. This technique was highly expensive due to repeated application and delayed effect. It also contaminated the environment, and through food chain, humans and their pets were affected. Baits and dusts were also used. Emulsifiable concentrate (EC) requires water for mixing, which is a disadvantage in remote and semiarid areas. Thus, in the absence of suitable barrier spray products, ULV formulations proved to be very efficient and most widely used method for controlling desert locust, and its guidelines have been issued by FAO (Symmons 1993).

Nowadays, Global Positioning Systems (GPS) are comparatively leading to new improved navigation technology for aircraft and ground team for locust control operations. Due to accuracy of position of locust situation and status, using radio and wireless for calling aircrafts for spray targets becomes more valuable tool. With improved operator protection, training, protective clothing and other adequate facilities will also definitely enhance the control measures. FAO has launched eLocust2 and Reconnaissance and Management System of the Environment of *Schistocerca* (RAMSES), for obtaining data on preliminary survey regarding the status and location of locusts. The management and analysis of data can be quickly done by concerned national organisation using computers with GIS tools. Another system known as *Schistocerca* Warning and Management System (SWARMS) provides reports on weather and remote sensing obtained from other computer systems (Joffe 1995). SWARMS also reveals the specific local information based on analysis of data received from various sources, which can be displayed on a map with variable scales. Thus, we can have a thorough knowledge regarding the specific status of locust swarms along with the weather conditions in a specified area. Mathematical models have also been developed to provide prediction of swarm-forming locust population (Joffe 1995).

Consistent Monitoring of Environment After Locust Spraying
The spraying campaign against locusts showed some nontarget effects. Gruys (1992) reported the locust plague during 1986–1989; some nontarget organisms also died. Many other experimental studies were conducted on locust and grasshopper spraying in Mali by American and Norwegian scientists (Dynamac 1988; Johannessen 1991; Otteson et al. 1989, and Fiskvatn 1993). In another ecotoxicological research programme in Senegal, the level of pesticide residues from locust control operations and subsequent assessment of their effects on nontarget organisms was performed by Gruys (1992). The problem for environment impact assessment studies after actual locust control operations needs attention. Efforts are made by FAO to develop monitoring techniques for environment impact assessment.

The use of satellite imaging and forecast efforts are on to initiate satellite imaging system for estimating rainfall, other weather changes and vegetation indices in order to forecast locust occurrence and warning system.

Difficulty in Proper Information and Data Collection
Due to security reasons also, many affected countries and organisations conceal the factual information regarding locust hoppers and adults using satellite-based communication. An alternative to this is to use portable telephones, modem and other handheld gadgets for data collection and locust monitoring for an effective locust control strategy. Improved locust surveillance, monitoring and control of locust hoppers and swarming adults depend a lot on better communication of information to regional and national plant protection services. Certain points to keep in mind are:

(i) To access across all cross-border affected areas
(ii) Accurate field reporting from various sites, which are shifted with locust activity
(iii) Availability of low-cost information technology
(iv) Better sharing of data relevant to locust control at international level

Alternative Control Methods

Other methods in addition to existing strategies for locust control may be:

(i) *Control by natural enemies* (Table 1.5): In nature, there are many enemies of *S. gregaria* and other locusts and grasshoppers including vertebrate (amphibians, reptiles and birds), invertebrate predators, parasites (mites, protozoans and nematodes), parasitoids (Hymenopterans) (Fig. 1.36) and pathogens (Fungi). There are certain mites belonging to genera *Eutrombium* sp. and *Caloglyphus* sp., which suck the contents of eggs and hatching is hampered. Entomopathogenic nematodes play an important role in insect control. Most important species of such nematode is *Heterorhabditis bacteriophora.* The nematode larvae and adults enter the insect body through spiracles. The nematodes harbour some bacteria as symbiont, which help in killing the insect hosts. The pathogens used in insect pest control are fungi, bacteria and viruses. The potent fungi that may be used in insect control is *Metarhizium anisopliae var. acridum* (Figs. 1.37 and 1.38) and *Beauveria bassiana. Beauveria bassiana* causes muscardine disease in insects. They reproduce by conidia and form a fungal mat on the outer surface of insect body. *Bacillus thuringiensis* and NPV are important pathogens that can be promising biological control agent for the management of locust outbreaks. The natural enemies are able to control locusts up to some extent only, as they are unable to follow locust swarms in the next breeding area (Uvarov 1977). They cannot prevent upsurges but can hasten the termination of plague occurrence (Greathead 1992).
(ii) *Antifeedants*: As the name indicates, these chemicals inhibit the gustatory sensilla, preventing them from feeding. The extract of neem may be used as an antifeedant, which may cause some abnormalities during development.
(iii) *Juvenile hormone analogues (JHAs):* The use of juvenile hormone analogues or mimics may disrupt the sexual maturation of locusts. Without sexual maturation, further reproduction is not possible.
(iv) *Pheromones*: Also known as ectohormones, they are secreted by either sex in insects. They modify the behaviour of the recipient. Efforts are on to investigate their potential in modifying behaviour and development with an aim to disrupt swarm formation (Raina 1992).
(v) *Semiochemicals*: The chemicals regulating insect behaviour are collectively known as semiochemicals. The insect behaviour could be modified (exploited) to the advantage of man. They may be of plant or insect origin or synthetic analogues, which aim at studying the behaviour regulating chemicals and exploiting them for insect pest control. In relation to locusts also, the scientists

Table 1.5 List of natural enemies of desert locust

S. No.	Natural enemy	Class	Type	Life stages
1	*Acanthodactylus cantoris*	Lizard	Predator	Adults/nymphs
2	*Argiope arcuata*	Spider	Predator	Adults/nymphs
3	*Bacillus thuringiensis*	Bacteria	Pathogen	
4	*Beauveria bassiana*	Fungus	Pathogen	
5	*Blaesoxipha agrestis*	Fly	Parasite	Adults/nymphs
6	*Blaesoxipha filipjevi*	Fly	Parasite	Adults/nymphs
7	*Gregarina garnhami*	Unicellular organism	Pathogen	Adults/nymphs
8	*Gregarina ridiga*	Unicellular organism	Pathogen	Adults/nymphs
9	*Lysinibacillus sphaericus*	Bacteria	Pathogen	Adults/nymphs
10	*Malamoeba locustae*	Protozoa	Pathogen	Adults/nymphs
11	*Mantis religiosa*	Insect	Predator	Adults/nymphs
12	*Metarhizium anisopliae*	Fungus	Pathogen	Adults/nymphs
13	*Metarhizium flavoviride*	Fungus	Pathogen	Adults/nymphs
14	*Nosema locustae*	Bacteria	Pathogen	Adults/nymphs
15	*Nucleopolyhedrosis virus*	Virus	Pathogen	Adults/nymphs
16	*Passer domesticus*	Bird	Predator	Adults/nymphs
17	*Pseudomonas aeruginosa*	Bacteria	Pathogen	Adults/nymphs
18	*Schizodactylus monstrosus*	Insect	Predator	Adults/nymphs
19	*Seniorwhitea krameri*	Fly	Parasite	Adults/nymphs
20	*Sphex nivosus*	Wasp	Predator	Adults/nymphs
21	*Steinernema carpocapsae*	Nematode	Parasite	Adults/nymphs
22	*Stomorhina lunata*	Fly	Parasite	Eggs
23	*Symmictus costatus*	Fly	Parasite	Adults/nymphs
24	*Systoechus aurifacies*	Fly	Predator	Eggs
25	*Systoechus somali*	Fly	Predator	Eggs
26	*Trox procerus*	Beetle	Predator	Eggs
27	*Uromastix hardwickii*	Lizard	Predator	Adults/nymphs
28	*Eutrombium* sp.	Mite	Parasite	Eggs
29	*Caloglyphus* sp.	Mite	Parasite	Eggs

Source: CABI

Fig. 1.36 *Scelio* spp. parasitic wasps (Hymenoptera) hatching from acridid eggs. (Source: CABI, datasheet)

Fig. 1.37 Cadaver of *S. gregaria* killed by *Metarhizium anisopliae* var. Acridum. (Source: CABI)

Fig. 1.38 Disarticulated cadaver of *Z. variegatus*, killed by the fungus *Metarhizium anisopliae* var. *acridum*; green spores are visible inside. (Source: CABI, datasheet)

working in this field should explore the possibility for locust control using this method.

(vi) *Insect growth regulators* (IGRs): This is a class of insecticides which disrupt the moulting process in insects, and they are unable to develop into adults. Chitin synthesis inhibitors, such as diflubenzuron, are a chemical which prevents the synthesis of chitin in developing insects. Hopper stage is the most vulnerable stage of locusts. This chemical is spread as barrier treatment in rows of marching hoppers. This substance is absorbed by the immature stages, and they die during moulting into next stage. Extra care and vigilance is needed to detect the right time for using IGR before wingless hoppers become winged adult and make swarm. Insect growth regulators have not been tested on large-scale operational trials, although they have opened the door to a new locust control strategy. IGR is persistent on the treated vegetation but exhibits very low toxicity to vertebrates; hence, their use is recommended.

Besides using all these methods for locust control strategies in a coordinated manner and with the blooming technologies, such as the satellites, high-tech computers and mathematical models, at our disposal, we would be able to restrict the damage caused by locusts.

1.7 Conclusions

The locusts are grasshoppers that belong to order Orthoptera, suborder Caelifera and family Acrididae of class Insecta. The suborder Caelifera contains about 12,000 species, out of which about 10,000 species belong to family Acrididae alone. It includes short-horned grasshoppers and locusts. The antennae are shorter than the body, and less than 30 segmented, tympanal organs are located on the first abdominal segment.

A locust is a migratory grasshopper that swarms at regular intervals. The periodicity of locust swarm is largely governed by complex environmental factors. The locusts exhibit polymorphism, as they occur in more than one form during adult life. According to the phase theory of locusts proposed by Uvarov (1966), the locust species exist in two main forms or phases, the gregarious phase (phasis gregaria) and solitary phase (phasis solitaria). The two extreme forms of solitaria and gregaria are connected by intermediate forms termed transiens. The gregarious forms make large swarms, which migrate over wide geographical regions and cause great devastation to natural and cultivated vegetation, forests, pastures and crops. In ancient periods, they were, therefore, called the plague, meaning 'curse of God'.

The desert locust, *S. gregaria*, is the most destructive species of locust in Indian subcontinent causing severe damage while migrating from Northwest Africa, through Middle East to Indian subcontinental countries and Portugal, Turkey, Uzbekistan and Tanzania. The breeding of locusts generally coincides with the rain fall. The eggs are laid in moist soil in the transitional area between the arid and semiarid regions.

The life history of locusts passes through three stages, namely, egg, nymph and adults. After a total duration of about 3–10 weeks of nymphal stage, the adults emerge after final moult. Thereafter, morphological and behavioural changes occur in the hoppers, as they enter either the solitary or gregarious phase depending on the breeding in a scattered or concentrated manner, respectively. The gregarious adults migrate as swarms. At an international level, various organisations are engaged in locust control, such as:

(i) The Locust Warning Organization (LWO) set up in 1939 in India
(ii) Anti-locust Research Centre (ALRC) located at London
(iii) Field Station of Investigation on Locusts (FSIL) located in Bikaner (Rajasthan, India)
(iv) Anti-locust Operation under Food and Agricultural Organization of United Nations (FAO)

The organisations alert the persons concerned about outbreaks and swarms of locusts. In India, very recently, the swarms of desert locust have invaded most of the areas of Rajasthan. A continuous and consistent vigilance and field survey on the hopper populations in the breeding areas, using modern methods for sending information and using all possible methods of control will definitely pose a check on locust problem.

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Termites

2

Mani Chellappan and M. T. Ranjith

Contents

Abstract

Termites are eusocial insects belonging to the insect infraorder Isoptera and are characterized by their colonial behavior. The word Isoptera originated from the Greek words *isos* which means equal and *pteron* which means wing and refers to the two pairs of identical wings in the adult. Termites are polymorphic insects, living in large communities of several hundred to several million individuals, composed of alate or apterous reproductive forms together with numerous apterous sterile soldiers and workers. Their numerous colonies have great influence in many ecosystems. There are 12 families of which the family Termitidae is the largest accounting about 75 percent of all termites. With the peculiar digestive system and the ability to digest lignocelluloses, the most abundant resource on the planet, termites became the most successful insects. Termites built huge and most

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M. Chellappan (✉) · M. T. Ranjith
AINPVPM, Department of Agricultural Entomology, College of Horticulture, KAU, Thrissur, Kerala, India

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complex nesting systems ever known by an insect. They change the ecosystems by their activities, and at the same time, they are dreaded pests on agriculture and man-made wooden structures. Due to their cryptic life, it is very difficult to manage them. Though chemical insecticides are very effective on termites, their method of application is challenging. Killing few thousand termite workers does not mean killing the colony; as long as the primary and secondary reproductives are alive and active deep inside the termite mounds, the termite problem exists, perennially.

Keywords

Termite · Mound · Pseudergates · Worker · Alates · Reproductives · Symbionts

2.1 Introduction

Termites are a fascinating, eusocial and polyphagous insects (Eggleton 2011; Narayanan 2011; Narayanan and Thomas 2016). Fossil record indicates that termites evolved about 220 million years ago (Collins 1988; Thorne and Carpenter 1992). They are small insects, but the combination of their sociality with their ability to efficiently digest lignocellulose led to a tremendous evolutionary success (Brune 2014). They are said to be derived from a primitive group of wood-dwelling cockroaches (Fig. 2.1), clearly seen in the obligate dependence on mutualistic intestinal protists and, in some higher forms, externally cultivated basidiomycete fungi (Bignell and Eggleton 1998). They live in a highly organized system of small

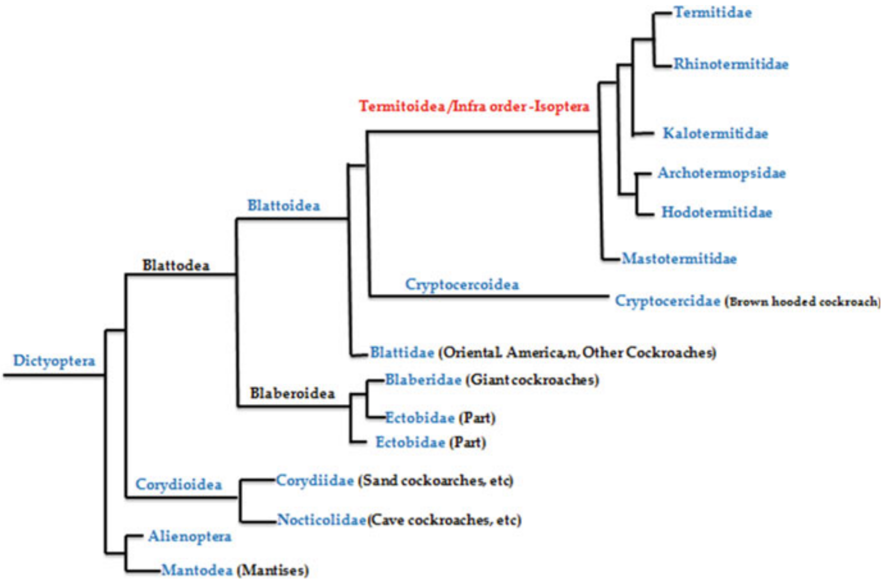

Fig. 2.1 Phylogeny of termite

and large communities in different ecosystems. There are approximately 1 X 10^{18} termites on earth, and they can comprise 10 percent of tropical animal biomass and 95 percent of the biomass of soil insects (Bignell 2006). Out of 3106 described termite species (Krishna et al. 2013), about 270 occur in agricultural fields and destroy crops (Rouland-Lefevre 2011) and 26 on wood (Evans 2011). Based on their dietary habits, they are generally divided into lower termites (which feed on wood like cockroaches) and higher termites (highly eusocial and have broader feeding habits) (Sun and Zhou 2012). Termite societies are characterized by the occurrence of polymorphic forms each assigned with specific functions, presence of individuals of several generations at the same time in a particular nest, and occurrence of worker caste which provides food and care for the nest members and with clear division of labor. As eusocial insects, individual termites cannot perform all the functions needed for survival, and the whole colony exists as a super organism (Eggleton 2011).

Termites are a keystone species in many ecosystems, especially in the semi-arid regions (Black and Okwakol 1997). They construct nests to maintain constant temperature, and humidity gives them the ability to remain active in hot, dry conditions (Jouquet et al. 2011). In hot areas, termites play an increasingly important role in improving soil structure, water-holding capacity, and fertility and undertake many of the soil development activities that earthworms provide in cooler and wetter areas (Evans et al. 2011). In addition to termites' impact on ecosystems, they have also been used in scientific research, medicine, and technology (Bignell 2006).

In natural ecosystems, termites are beneficial creatures, breaking down cellulose material, such as decaying plant material, decomposing trees, and leaf litter (Table 2.1). Termites become a problem in urban areas (building materials and structures made up of wood products) and in cropped land, where numerous crops plants are damaged by these cryptic and soil-dwelling insects (Green 2018). In this chapter, the diversity, distribution, positive and negative impacts of termites on ecosystems and human activities, integrated management methods, etc. are being examined.

2.2 Diversity and Distribution

The word Isoptera originated from the Greek words *isos* which means equal and *pteron* which means wing and refers to the two pairs of identical wings in the adult (Harris 1957; Thorne and Carpenter 1992). Classified in 12 families based on the morphology and the associated symbionts, there are about 3106 living and fossil termite species in the world (Krishna et al. 2013), and these are grouped as follows.

2.2.1 Systematic Classification of Termites

Kingdom: Animalia
Phylum: Arthropoda

Table 2.1 Termite families and feeding groups

Feeding group	Termite groups
Group I Lower termite - Deadwood and grass feeders.	Hodotermitidae Kalotermitidae Mastotermitidae Rhinotermitidae Serritermitidae Termopsidae
Group II Higher termite - Deadwood, grass, leaf litter, and micro-epiphytes.	Amitermitinae (part) Cylindrotermitinae Protohamitermitinae Macrotermitinae *Nasutitermes* group Nasutitermitinae (part) Sphaerotermitinae
Group III Higher termite - Organic rich, upper layer of soil.	*Amitermes* group (part) *Pericapritermes* group (part) *Anoplotermes* group Apicotermitinae (part) Foraminitermitinae *Nasutitermes* group Nasutitermitinae (part) Subulitermes group *Termes* group (part)
Group IV Higher termite - True soil feeding .	*Amitermes* group Amitermitinae (part) *Anoplotermes* group *Apicotermes* group Apicotermitinae (part) Apicotermitinae (part) *Cubitermes* group Nasutitermitinae (part) *Pericapritermes* group (part) *Subulitermes* group

Class: Insecta
Subclass: Pterygota
Infraclass: Neoptera
Superorder: Dictyoptera
Order: Blattodea
Infraorder: Isoptera
  Lower termites
  Family Cratomastotermitidae
  Family Mastotermitidae
  Family Termopsidae
  Family Archotermopsidae
  Family Hodotermitidae
  Family Stolotermitidae
  Family Kalotermitidae

Higher termites.
Family Archeorhinotermitidae
Family Stylotermitidae
Family Rhinotermitidae
Family Serritermitidae
Family Termitidae

2.2.2 Termite Family: Termites are Grouped Under 12 Major Families

2.2.2.1 Family Cratomastotermitidae

This has only one genus, *Cratomastotermes*, which is extinct, now.

2.2.2.2 Family Mastotermitidae

This family is the most primitive of all the termite species and found (*Mastotermes darwiniensis*) only in Northern Australia and Papua New Guinea. The fossils (13 fossil records) found of this species worldwide date back to the Cenozoic period (Encyclopedia 2020). The hindwings of Mastotermitidae are very similar to those of the cockroach, and the females of the *Mastotermes* lay egg cases that have up to 24 eggs arranged in two regular rows. In the colony, there are many secondary reproductives that raise the populations, while the primary reproductives could hardly be seen. Mastotermitidae soldiers are many, and the soldiers have powerful mandibles and excrete a toxic brown substance to repel intruders.

2.2.2.3 Family Termopsidae

Members are commonly called as damp-wood termites. They are arguably the largest termites in the world. They come in at a whopping 25 mm in length. There are around 20 species in the world, spread among the Americas, Eurasia, Africa, and Australia (Encyclopedia 2020). In contrast to the body size, their colony sizes are among the smallest. For instance, in the Pacific damp-wood termite (*Zootermopsis angusticollis*), the reproductive adults are around 23–26 mm in size including wings. The soldiers are around 15–20 mm in size. Workers are around 10–15 mm in size. Workers are pale milky white in color. Soldiers are darker pale in color with a dark brown head region. The queens of damp-wood termite species produce around 30 or less a day, and the colony sizes are around a moderate 10,000 members on an average.

Damp-wood termites are generally brown in color but can range from dark to light and can even be reddish, especially toward their abdomens. Nymphs are cream-colored. A characteristic feature of these termites is this species needs regular contact with water and a high humidity to survive.

Damp-wood termites do not create shelter tubes, as other subterranean termites do, and can live completely within the wood that they are feeding. These termites plug openings into the wood they are infesting with their fecal pellets to preserve humidity, and their cryptic behavior can keep them from being discovered for years. In nature, they live in the forests and nest in the wet, rotting, and rotten woods on the forest floor. Damp-wood termites usually infest felled timber, dead trees, and stumps and then feast on the contents. They can infest structures where high moisture levels exist from plumbing leaks, ventilation deficiencies, or drainage problems. Even the old gutters filled with leaves can also cause excessive moisture to accumulate in wall cavities, creating an attractive environment for these humidity-loving insects. *Archotermopsis*, *Hodotermopsis* (Eurasia, temperate zone), *Zootermopsis* (North America), *Stolotermes* (Africa, Australia, New Zealand), and *Porotermes* (Africa, Australia, New World – Chile only) are few genus under this family (Wilson 1971; Thorne and Haverty 1989; Abe and Higashi 2001).

2.2.2.4 Family Archotermopsidae

Members are also commonly called as damp-wood termites and formerly included within the family Termopsidae. This is a small family with five extant genera which may be a nuisance rather causing extensive damage to buildings or other structures; they eat wood that is not dried out or rotting.

2.2.2.5 Family Hodotermitidae

The Hodotermitidae family is known as the grass-harvesting termites. This family currently has 15 species that closely resemble the damp-wood termite species. The grass-harvesting termites are fairly large, at around 15 mm in size. This species occurs most commonly in the grassy lands of India, the Middle East, and Africa.

2.2.2.6 Family Stolotermitidae

Stolotermitidae comprise ten living species in Australia, sub-Saharan Africa, and the Neotropics (Emerson 1942; Krishna et al. 2013). The family has historically been included in the Termopsidae. Strictly wood-dwelling termite, members of Stolotermitidae nest in the decaying logs and have no true worker caste in the colony.

2.2.2.7 Family Kalotermitidae

This is the dry-wood termite family. The name dry-wood comes from the family's need to feed on wood above the ground, unlike most termites who mostly come into contact with the wood through the soil. However, this species has habits to feed and dwell in damp wood and rotten wood. There are over 400 species of dry-wood termites worldwide. This family is also found commonly in almost all continents with the exception of Antarctica.

2.2.2.8 Family Archeorhinotermitidae

Archeorhinotermes is the sole genus of the family and now extinct (Krishna and Grimaldi 2003).

2.2.2.9 Family Stylotermitidae

This contains one extant (*Stylotermes*) and two extinct (*Parastylotermes* and *Prostulotermes*) genera.

2.2.2.10 Family Rhinotermitidae

The family Rhinotermitidae is better known as the subterranean termite family and is known for its typical requirement of soil contact for nest construction. However, there are some exceptions in some genera. In Rhinotermitidae, the pronotum is flat and crescent shaped; typically, the width is less than or equal to the width of the head. The soldiers of this family are known for having the pronotum and for producing a defensive fluid. Both the workers and soldiers of this family are quite small, less than 5 mm, and the winged termites or reproductive adults are around 10 mm in length in eastern subterranean termite (*Reticulitermes flavipes*). However, in Formosan subterranean termite (*Coptotermes formosanus*), the soldiers are a pale yellow to pale tan color with oval-shaped heads and sickle-shaped mandibles. The workers are a pale yellowish brown and lack enlarged mandibles. Reproductive males and females are a yellowish brown with hairy wings. The termites with wings, or reproductive adults, are around 12 to 17 mm including their wings. The workers and soldiers are from 6 to 19 mm in size. The average queen in this family produces 100 termite eggs a day. This family is found on every continent and lives under damp conditions except for the polar regions and nearby lands.

There are about 158 extant species and 13 fossil species of Rhinotermitidae. There are more than 300 species worldwide under different genera, viz., *Psammotermes*, *Coptotermes*, *Heterotermes*, *Reticulitermes*, *Prorhinotermes, Termitogeton*, *Parrhinotermes*, *Schedorhinotermes*, and *Rhinotermes* (Encyclopedia 2020; Wilson 1971; Abe and Higashi 2001; Chouvenc et al. 2016). Genus *Reticulitermes* is widely distributed in North America and other temperate and subtemperate regions and a serious pest; *Coptotermes* is a serious pest on crops in tropical and subtropical regions.

2.2.2.11 Family Serritermitidae

The Serritermitidae family is very similar to the family Rhinotermitidae, which are the subterranean termites. They prefer to create nests with contact to the soil. There is a single species in the world, and this species occurs only in South America. The soldiers have unique mandibles that have serrated teeth-like projections on the inside.

2.2.2.12 Family Termitidae

Termites of this family come under the group called as higher termites. Termitidae is the largest termite family (about 75% of all termites) of which 2100 are living and three are fossil species and distributed widely (Table 2.2). The subfamilies, viz., Apicotermitinae, Termitinae, Macrotermitinae, and Nasutitermitinae, vary in their morphology, social organization, and nesting habits. The members of this family are actually a beneficial species to the desert ecosystem. The family contains the builders of great mounds some recorded as high as eight meters. The species range is grouped

Table 2.2 Distribution of family Termitidae

Family Termitidae	
Subfamily Apicotermitinae	Distribution
Speculitermes	Asia, New World tropics
Anoplotermes	Africa, New World
Euhamitermes, Eurytermes, Indotermes, Protohamitermes	Asia
Apicotermes, Hoplognathotermes, Lepidotermes Trichotermes	Old World tropics
Ahamitermes, Incolitermes	Australia
Microcerotermes	Worldwide except for North America
Amitermes	Worldwide
Drepanotermes	Australia
Gnathamitermes	North America, temperate
Subfamily Termitinae	
Basidentitermes, Crenetermes, Cubitermes, Euchilotermes, Fastigitermes, Foraminitermes, Megagnathotermes, Noditermes, Ophiotermes, Pericapritermes, Procubitermes, Thoracotermes, Promirotermes, Thoracotermes, Unguitermes	Old World tropics
Cavitermes, Dentispicotermes, Neocapritermes, Orthognathotermes, Spicotermes, Spinitermes	New World tropics
Termes	Worldwide, in tropics
Angulitermes	Eurasia, Africa
Dicuspiditermes, Homallotermes, Microcapritermes, Procapritermes	Asia
Capritermes	Madagascar, Africa
Subfamily Macrotermitinae	
Acanthotermes, Allodontermes, Ancistrotermes, Protermes, Pseudocanthotermes, Sphaerotermes, Synacanthotermes	Africa
Macrotermes, Microtermes, Odontotermes	Asia, Africa
Subfamily Nasutitermitinae	
Eutermellus, Mimeutermes, Verrucositermes	Africa
Bulbitermes, Hirtitermes, Hospitalitermes, Lacessititermes	Asia, New Guinea
Grallatotermes, Trinervitermes	Asia, New Guinea, Africa
Armitermes, Constrictotermes, Convexitermes Cornitermes, Curvitermes, Labiotermes, Paracornitermes, Procornitermes, Rhynchotermes, Subulitermes, Syntermes, Velocitermes	New World tropics
Tenuirostritermes	New World
Nasutitermes	Worldwide, in tropics

around the tropical climates in Asia, Africa, Australia, and South America. Few species of this family are in the deserts of North America. There are over 1800 species in Africa alone. This species is known to be one of the most prolific animals in the animal kingdom, generating over ten million eggs a year per queen (Engel et al. 2009; Encyclopedia 2020; Wilson 1971; Abe and Higashi 2001; Eggleton and Tayasu 2001).

Unlike the lower termites, family Termitidae have lost the symbiotic protozoan (flagellates) associated with other termites. Instead, they possess a highly compartmentalized gut and additionally a highly diverse gut microbiome composed of bacteria and archaea. It is thought that the loss of the symbiotic flagellates allowed this group to exploit new niches and therefore the immense diversity. Members of Termitidae are known to feed on lichens, soil, humus, leaf litter, grass, various roots, and wood, inquilines feeding off of the nest material of other termites and even opportunistic carrion feeding. The subfamily Macrotermitinae has evolved to cultivate fungus to digest lignocelluloses for them (VanDyk 2020). Among the many species of this family, the desert termite, *Gnathamitermes perplexus,* is an important species that recycles the nutrients back into the earth.

2.2.3 Geographical Distribution of Termites

Major families and subfamilies of termite are distributed in all continents except Antarctica; due to their soft cuticles, termites do not inhabit cool or cold habitats (Sanderson 1996). The subfamily Macrotermitinae occurs in many tropical and subtropical regions, e.g., Africa, Arabia, Afghanistan, India, Southern China, Southeast Asian islands, and other countries except Australia and South America. Australia is populated by *Amitermes* and *Coptotermes,* which have evolved and spread widely in this continent (Harris 1964). In South America, Nasutitermitinae, Amitermitinae, and Termitinae are well represented (Araujo 1970). The neotropical regions of North America and some of the European countries are, however, inhabited mostly by lowly evolved termite families, e.g., Kalotermitidae, Hodotermitidae, and Rhinotermitidae. A few genera of the family Termitidae are also found occurring in these regions. Of the 3100 and odd termite species recognized currently, 1000 are found in Africa alone (Meyer et al. 1999).

2.3 External Morphology and Internal Anatomy

2.3.1 Size of the Body

Body size varies from small to medium in different castes and species of the termites. Usually, it ranges from 4 to 15 mm in length. For example, subterranean worker termites may be as short as 3.2 mm but grow to be up to 6.35 mm or maybe as much as 9.53 mm in length. The soldiers found along with these subterranean termites will be slightly longer, while the reproductive queens and kings will be over 19 mm. In case of Formosan subterranean termites, they are 10–20% larger than the common subterranean termites. However, the largest of all extant termites is the queen termite of *Macrotermes bellicosus,* which measures about 100 mm in length (Anna 2013).

2.3.2 Color of the Body

Body color of termites ranges from yellow, brown, black, or pale in different castes under different species. Usually, termites have weakly sclerotized and pale body. Worker termites often appear lighter, while swarming termites are darker. Soldiers tend to have worker-like bodies but with hard heads that are often dark in color. Alates (reproductives/swarmers) have hard exoskeletons that may be very dark. There are variations in color between termite species. For example, western subterranean termite soldiers have yellowish heads, while western dry-wood termite soldiers have reddish brown heads.

2.3.3 Body Structure

Termite body has distinct three regions, viz., the head, thorax, and abdomen. The termite head is generally prognathous, and the shape of the head capsule varies dramatically between soldiers and the worker-reproductive castes. Soldiers have a longer, rectangular-shaped head, except for nasute soldiers (Termitidae: Nasutitermitinae) where it is bulbous. The soldiers of some Kalotermitidae have plug-shaped (phragmotic) heads (Krishna et al. 2013), which are employed to close the openings to the nest tunnels. Though the head bears a pair of compound eyes, they are functionless (except in reproductives). A pair of moniliform antennae with varying number of distinctive moniliform segments and the biting and chewing mouthparts with well-developed mandibles are the other appendages present on the head. Termites have three prominent mouthparts, viz., the labium, the maxillae, and the mandibles. The labium and maxillae are used to sense and handle food. Mandibles are simple in reproductives, whereas the soldiers have enlarged defensive mandibles. Worker mandibles are either heavily sclerotized for grinding plant material or work together like a mortar and pestle for pounding soil or humus (Donovan et al. 2001: Eggleton 2011). The neck has a prominent lateral sclerites, and the body has a prominent pronotum. Except the reproductives, no other caste possesses wings. On alates, a pair of equal wings (*iso – ptera*) arises from meso- and metathorax segments. The legs are cursorial with four segmented tarsi with a pair of claws. Termites have a ten-segmented abdomen. The tenth abdominal segment ends in a pair of short two- to six-jointed cerci; in male, the ninth segment has a pair of anal styles. The reproductive organs of termites are simple but closely resemble to those in cockroaches (Nalepa and Lenz 2000). Sternal glands are present within the intersegmental membrane along the midline of the abdomen of worker termites. Secretions of the sternal glands are important in trail marking while in foraging activities (Krishna et al. 2013).

2.3.4 Internal Anatomy

Consistent with the members of class Insecta, termites share the basic internal anatomy and different systems as that of other insects. The digestive system consists of the typical three divisions found as in other insects: the foregut, midgut, and hindgut. Food gets mixed with saliva and stored in the crop (Wigglesworth 1972). Thick muscle layers are present in the crop, proventriculus and rectum; in the proventriculus, the thick muscle layer together with six dentitions aids in grinding the food. The hindgut, or proctodeum, is special in termites and is divided into five segments (Fig. 2.2): first proctodeal segment followed by the enteric valve, paunch, colon, and rectum (Krishna et al. 2013). The paunch is dilated and voluminous and harbors symbiotic protozoans in lower termites or the symbiotic bacteria in the Termitidae. Protists that reside in the termite gut ingest wood particles in the form of cellulose and degrade it within their cells. Cellulolytic protists known as *Trichonympha* and *Mixotricha* produce cellulases and various glycolytic enzymes that can break down cellulose (Abe et al. 2000).

2.3.5 Termite Feeding

Almost all species of termite are detritivorous. They consume wide range of freshly dead or decaying plant material including dry grass, leaf litter, decaying wood, dung, humus, carpet, cardboard, insulation, sheetrock, paper, fabric, animal excreta, and

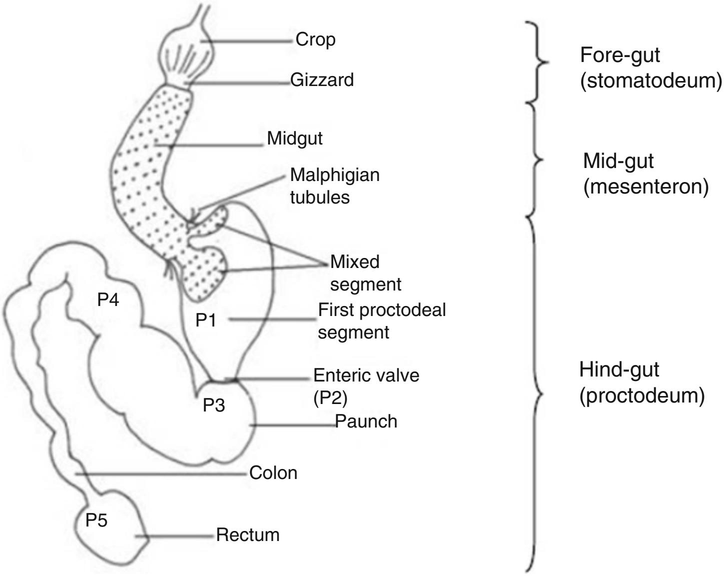

Fig. 2.2 Termite gut

carrion. Living plant tissues, including lichens and mosses, are taken by a few species. Another feeding group that may be common and important in many tropical forests is the soil-feeding termites. Accurate information on the natural history and feeding habits of termites is still scarce for some groups, particularly the subterranean species. Nonetheless, termite species can be categorized into five broad trophic categories according to their food, foraging galleries or columns, color of the abdomen, and known biology (Eggleton et al. 1997). In nature, termites are helpful because they are able to break down decaying trees and dying plants. However, the digestion of cellulose is not an easy task. Even large animals, such as ruminants, have difficulty stomaching the substance. To get past that, those animals chew grass for long periods of time until the cellulose is more easily digested. Termites are able to live off cellulose owing to the organisms found in their stomachs. Bacteria and protozoa form a mutually beneficial relationship with the pests by producing a special enzyme that naturally breaks down cellulose. The microbes digest the cellulose, and termites receive their nutrition in the form of sugar. Additionally, some termite species favor wood that's already being broken down by fungi, like *Termitomyces* sp., to make digestion easier. Immature termites that do not yet have the bacteria and protozoa in their stomachs, soldiers, and reproductives are fed by workers. Worker termites pass on the cellulose turned sugar substance via a mouth-to-mouth feeding process. Evolution of feeding habits in termites showed that many families have evolved into soil feeders only to later revert to wood or humus feeders (Table. 2.1) and that this has occurred on several occasions (Donovan et al. 2001; Cuttiford 2011).

2.3.6 Trophic Groups

2.3.6.1 Wood Feeders

These primitive wood-eating termites feed on wood and woody litter, including dead branches still attached to trees, and they may live in their feeding galleries, which in some cases become colony centers (Eggleton et al. 1997). The condition of wood taken is very important. This may include living trees (*Coptotermes*, *Schedorhinotermes*, and *Microcerotermes dubius*), sound dead wood (Kalotermitidae), and/or fungus-attacked wood (Nasutitermitinae, some Termitinae, and Macrotermitinae). Most of these termites are arboreal (attached to trees), subterranean, or epigeal nesters (Eggleton et al. 1997).

2.3.6.2 Soil Feeders

These are termites feeding on the upper mineral soil. The vast majority of species in this group ingest top soil rich in organic matter. They are normally distributed in the soil profile, in the organic litter layer (plant debris), and/or in epigeal mounds (Eggleton et al. 1997). This form is found in many Termitinae and most Apicotermitinae (Eggleton et al. 1997; Abe et al. 2000). Soil feeders are very common and abundant in many tropical rain forests.

2.3.6.3 Soil-wood Interface Feeders

Termites in this group feed on highly decayed (friable and soil-like) wood, soil under logs or soil plastered to logs, or soil mixed with leaf litter in stilt-root complexes (Eggleton et al. 1997). Soil-wood interface feeders are only found in Termitinae, Apicotermitinae, and Nasutitermitinae. Most of them nest within dead logs, build epigeal nest, or form colony centers in the soil.

2.3.6.4 Litter Foragers

These are termites that forage for leaf litter and small woody item litter in various stages of decay. Food sources are often taken back and stored temporarily in the nest. This group includes some subterranean and other mound-building Macrotermitinae (with fungal association), as well as certain Nasutitermitinae that forage on the surface of the ground or litter layers (Eggleton et al. 1997). Genera *Laccessititermes* and *Longipeditermes* are also known as arboreal forages.

2.3.6.5 Micro-epiphyte Feeders

Termites of this group forage for moss, algae, lichens, and fungi on tree barks. Grass feeders will also take dung and may sometimes scavenge vertebrate corpses. Grass feeders are mainly of the family Hodotermitidae and found only in savanna and deserts (Krishna 1970).

2.3.7 Termite Symbionts

Termites and their microbial gut symbionts are major recyclers of lignocellulosic biomass. This important symbiosis is obligate but relatively open and more complex in comparison to other well-known insect symbioses (Rahman et al. 2015). Interaction between the microbes in the termite gut is highly mutual, usually beneficial for both microbes. Prokaryotes are closely associated with protists as symbionts, either attached to the cell surfaces or live within the cytoplasm or nucleus of the protists. For instance, *Treponema* spirochete bacteria are attached to the special bracket-like structures on the plasma membrane of *Mixotricha* and contribute to the movement of the host protist known as “motility symbiosis”. *Treponema* also benefit by living on and within the protist, easy access to nutrients H_2 and CO_2 produced by *Mixotricha* that they utilize to synthesize acetate and obtain energy for their own growth as well (Abe et al. 2000; Ohkuma 2001).

2.4 Social Structure

Termites, like other socially evolved insects such as honey bee and ants, are characterized by cooperation in the rearing of young individuals, sharing of resources (i.e., food, water, and shelter) among the individuals, overlapping of generations (i.e., year-round egg laying), and a division of labor among the individuals, which is characterized by the presence of one or more castes or life forms or polymorphism. Polymorphism is the occurrence of several forms within the same species where the individuals at the center of the colony develop gonads and reproduce sexually. Individuals at the periphery expose themselves to danger of combat and do not reproduce sexually.

Termites generally live in closely associated societies called colonies. A colony is a collection of individuals that cooperate in the rearing of young ones and that share resources (e.g., food and shelter) among themselves. Termite colonies are composed of a few adult forms (the king and queen), while the majority of the population consists of immature forms that are equal in numbers of males and females. Most of the social insects have a colony structure wherein the majority of members are females that live in a single, centralized, immobile nest from which workers forage in search of food and water, whereas termite social groups are composed of both males and females and have a mobile nest site usually located near food source, most often the dead wood, which they excavate and inhabit (Suiter et al. 2016).

2.4.1 Termite Castes

Termites exhibit a caste system of organization, wherein physically distinct individuals perform different tasks in the highly structured termite colony (Figs. 2.3 and 2.4). The caste of each individual is determined during its postembryonic development (Laine and Wright 2003) which is largely dependent

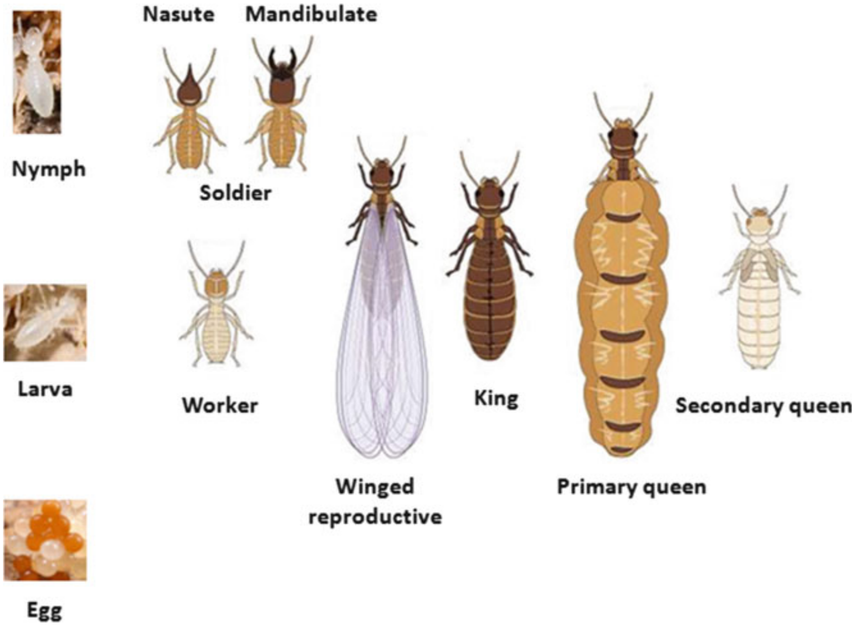

Fig. 2.3 Termite life stages and castes

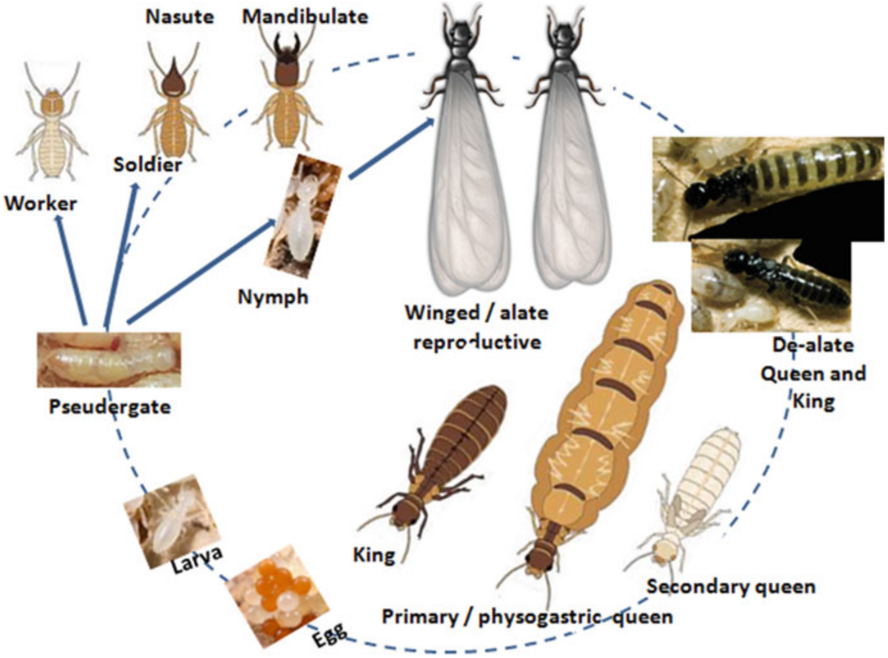

Fig. 2.4 Termite life history

on the pheromones present in the colony. The caste system in termites can vary according to type of species (Roisin 2000). In family Termitidae, the bifurcation occurs after the first stage of development, where an apterous line gives rise to the worker caste, after one or two larval instars. Subsequently, the worker caste gives rise to soldiers. A nymph line, after five nymphal instars with wing buds, gives rise to alates or reproductives (Roisin 1996; Moura et al. 2011).

Termites have four castes resulting from the expression of alternative postembryonic developmental trajectories. The first two types of castes are worker castes, which include both (1) true workers and (2) pseudergates or false workers. True workers are the individuals diverging early and irreversibly from the imaginal development (Lima et al. 2013b). The pseudergates are the individuals separating late from the imaginal line (Noirot and Pasteels 1987). The third caste is alates or imagoes or reproductives, which develop normally as other hemimetabolous insects. The fourth caste is soldiers, which are formed from a pre-soldier or white soldier stage after the postembryonic developmental pathway (Noirot and Pasteels 1987).

2.4.1.1 Worker Caste

Worker termites are physically and sexually immature males and females and are the most numerous caste in a colony. They are functionally sterile and are responsible for all the labor required in the nest. As their name suggests, they do most of the work in the colony. Workers of lower termites are typically with unpigmented and white or black body and blind and show no signs of wing development. They are soft-bodied and possess hard mouthparts, which are especially adapted for chewing of wood. They have smaller, saw-toothed mandibles, which allow them to take small bites of wood and carry building materials. The young workers attend colony duties like feeding, grooming, and caring the young ones, while the older workers are recruited on the hazardous jobs of foraging and nest building. They digest cellulose in ingested food and feed other nest mates by a process known as trophallaxis. Trophallaxis is an effective nutritional tactic to convert and recycle nitrogenous components. In *Macrotermes bellicosus* (Smeathman), the feeding habit differ in old and young workers, also referred to as major and minor workers, respectively. Major workers are foragers and main food processors, while minor workers attend to the queen. It is also found in the same species that as age increases, it may result in a change of performance in certain workers, from queen caring to external foraging activities (Hinze and Leuthold 1999).

2.4.1.2 Pseudergates or False Worker Caste

Sterile workers are sometimes termed as true workers, while those that are fertile, as in the wood-nesting Archotermopsidae, are termed as false workers. Pseudergates are false workers, which perform duties of workers like food collection, construction, or brood care but still possess the capacity to become reproductive (Roisin and Korb 2011). Grasse and Noirot (1947) coined the term “pseudergate” or “false worker” to designate the individuals who pass through regressive molts in *Kalotermes flavicollis* (Fab.). Generally, the apterous line in termites develops into workers and soldiers. Typically, the wood-dwelling termite caste patterns could be divided into two groups, one characterized by the development of pseudergates and the other by an early developmental fork, separating nymphal-alate line from the apterous line (Roisin and Korb 2011). Pseudergates can even indulge in attack and defense apart from doing other household jobs, as in those of *Hodotermopsis sjostedti* Holmgren (Ishikawa and Muira 2012).

2.4.1.3 Soldier Caste

Soldier termites are physically and sexually immature males and females whose primary function is colony defense. Soldiers are easily identified by their enlarged, yellowish to yellowish-brown head, and long, hard black mandibles (mouthparts) enable them to defend the colony against many predators. Many species are readily identified using the characteristics of the soldier’s larger and darker head and large mandibles. Some species can secrete a toxic or sticky substance from their heads, which they use to kill or subdue intruders.

Soldiers cannot feed on their own and are dependent on fellow workers to be fed. The soldier termites are wingless, blind, and otherwise soft-bodied. Soldiers mature in a year and may live up to 5 years (Myles 2005). The proportion of soldiers in a termite colony is specific to each species (Haverty and Howard 1981) which is also dependent on environmental factors in which they thrive (Liu et al. 2005a, 2005b). This proportion might have been influenced by two levels of internal regulation (Korb et al. 2003). The first is the individual endocrinal regulation, where change in juvenile hormone titer activates the differentiation process (Mao et al. 2005; Park and Raina 2004), whereas the second is the social regulation, where the present soldier proportion becomes template for the production of new soldiers (Park and Raina 2005; Mao and Henderson 2010). In *Reticulitermes*, soldiers typically comprise 1–2 percent of the termites present in a colony, whereas subterranean termite soldiers comprise 10–15 percent of the population (Suiter et al. 2016). Different types of soldiers include minor and major soldiers and nasutes, which have a hornlike nozzle frontal projection (a nasus). In most species, soldiers have large heads and strong, pincerlike mandibles.

2.4.1.4 Reproductive Caste

Reproductive caste is divided into different types based on the sex, function, and appearance.

2.4.1.4.1 Alates

Adult winged termites are called alates or swarmers, and they have fully functional wings and eyes, and their pigmented, dark skin can better tolerate water loss than the abovementioned castes. They are young winged reproductives of both sexes. The sudden, dramatic appearance of alate termites is commonly referred to as swarming and when it occurs indoors is often one of the first signs of a structural infestation (Philip 2004). From time to time, about 100 to 1000 alates leave the colony for a mating and colonizing flight, also called “nuptial flight.” Alates commonly succumb to desiccation, predation, and other environmental factors. Swarming occurs mainly when outdoor temperatures are warm and usually only by mature colonies.

2.4.1.4.2 Dealates

These are the alates that cast their wings after the colonizing flight and successively turn into queens and kings. After mating, a pair settles down at a suitable site like a rotting scar on a tree to establish a new colony. Initially, only a few eggs are laid and brought up by a female dealate. As the number of individuals in the colony grows, more workers are available to help the young queen to care for the brood. After three to five years, the colony matures and attains pest status.

2.4.1.4.3 King and Queen

After nuptial flights, the queens instantly search for nesting sites, while the kings follow them, and when they find a moist crevice with wooden material, a royal chamber in the form of nest is created, and the queen starts to lays eggs (Su and Scheffrahn 2000). A pair that successfully establishes a new colony is called the king (male) and queen (female) of the colony, and they are also referred to as the primary (first form) reproductives because they have attained the adult stage and are physically and sexually mature. The queen of the colony is responsible for egg production for the colony. Unlike in ants, the king mates with her for life. Within the first few days or weeks after pairing, the queen lays the first batch of several dozen eggs; small numbers of eggs are laid intermittently thereafter. The eggs are yellowish-white in color, and incubation period ranges from 50 to 60 days. In the very early stages of the colony, the queen lays only 10–20 eggs, but with nearly 2000 ovarioles (Kerkut and Gilbert 1985), it can lay thousands of eggs a day when the colony is several years old. In some species, the physogastric queen may produce 3000 to 40,000 eggs a day (Thompson 2000; Kaib et al. 2001). The termite queen shows the longest life span and lives up to 25 years or more (Myles 2005).

2.4.1.4.4 Neotenic Reproductives

These are often found in termite colonies, to assist the queen in laying eggs, once her productivity decreases. When the queen has died or deteriorated, one of the neotenics takes her place, and so they are also known as secondary queens. Hence, the removal of a queen from her colony does not necessarily mean the end of the colony. Unlike primary reproductives, neotenic reproductives do not develop wings, compound eyes, or uniformly pigmented skin. The worker-derived neotenics are called ergatoid reproductives that are smaller than nymphoid reproductives and do not possess wing buds (Thorne 1996).

2.4.2 Termite Habitat

Termites are highly successful insects. They practice many habits to survive, and the termite species spans the entire world. They thrive in the warm moist lowlands and along coasts. Some species have adapted to colder temperatures allowing them to infest homes and other wood sources in the north of equator. Termite habitats are the heaviest in the tropic regions, subtropics, and warmer climate regions.

2.4.3 Termite and Moisture Requirements

Termite colonies require moisture for survival and colonize the places where adequate moisture is ensured. Termite-supportive environment includes soil, dead and decaying wood, flat roofs where dead leaves and moisture have been allowed to accumulate, leaking pipes, areas with no ventilation, and leaking or poorly maintained gutters.

2.4.4 Termite Nest

Activities and interactions of termite species with soil environment have defined and modified many ecosystems for ages. Their influence is largely through their foraging, construction of nests, galleries, soil sheetings, and mounds. Their associated symbiotic relationship with microbes depending on the species also influences the soil and contributes to soil rehabilitation and plant diversity (Ali et al. 2013). Termites move a fourth of a metric ton of dirt to build mounds that can reach 17 feet (5 meters) and higher (Lisa 2014).

Subterranean termites make strong and elaborate underground networks of galleries and tunnels. World's largest soil mounds are constructed by termites (*Syntermes dirus*) that have persisted for up to 4000 years and cover an estimated 2,30,000 km^2 (approximately the size of the United Kingdom) of seasonally dry tropical forest in a relatively undisturbed and climatically stable region of Northeast Brazil (Martin et al. 2018). The mounds can survive fires and floods, which are used for food storage and raising the young ones. These underground networks give the colony a place to live, and they can connect the colony directly to sources of food. If there's an obstacle, the termite workers build shelter tubes. Shelter tubes are usually about the diameter of a pencil, and they are made of soil glued together with saliva. Depending upon the weather, the worker digs deep into the soil in search of warmth, if the weather gets cold; if it gets hot, the colony moves to parts of the nest that are shaded by aboveground vegetation or other structures. To ensure successful ventilation in different environments, the termites construct their mounds in coordination with their surrounding conditions. *Macrotermes bellicosus* constructs thin-walled, cathedral-shaped mounds in open savannas, for example, but thick-walled, dome-shaped mounds in forested areas (Ockoa et al. 2019). The mounds are dome- or towerlike structures made from soil particles and termite excreta glued together by the worker termites. A typical mound has multiple chimneys and tubes that allow air to circulate through the structure. The royal chamber accommodates the king and queen, and the inner layers of the mound contain galleries in which the termites live and raise young ones (Noirot and Darlington 2000). Some in the subfamily Macrotermitinae use underground galleries to grow symbiotic fungi, *Termitomyces* sp.

2.4.4.1 Wood Nesters

Termites in this group live in or around standing trees or dead logs. Sometimes the dead wood is gradually replaced with wood carton, a woody substance with low nutrient concentrations and high levels of lignin, and other undigested components (Collins 1988). This includes the kalotermids (*Kalotermes, Glyptotermes*), some rhinotermids (*Schedorhinotermes, Parrhinotermes, Heterotermes*, and *Coptotermes*), and some Termitidae members, such as *Microcerotermes* and *Termes*.

2.4.4.2 Hypogeal or Subterranean Nesters

These are termites whose colony centers are below the ground without any indication of their presence (Wood and Johnson 1986). They use their feces or a mixture of feces and mineral soil in nest construction. The colony centers are often poorly defined and characterless, especially in the soldierless Apicotermitinae. But in some Macrotermitinae, *Apicotermes* and *Homallotermes*, a little internal structure or surface holes present together with their complex underground nests. This is to enable the foragers to forage on aboveground vegetation. This group also includes many species that are facultative secondary inhabitants of epigeal mounds, *Microcerotermes*, *Pericapritermes*, and soldierless Apicotermitinae (Eggleton et al. 1996).

2.4.4.3 Epigeal Mound Builders

These are termites whose colony centers are associated with living (free standing or tree buttresses) or dead vegetation aboveground (Jones 1990; Eggleton et al. 1996), commonly known as mound builders. The mounds are usually well characterized, often with very complex structures. Materials used for construction are of three main types: subsoil with relatively low organic content added with salivary secretion (*Macrotermes* and *Cornitermes*), wood carton (a mixture of feces and macerated wood with a high lignin content) (most wood-feeding termites), or a mixture of feces and organic-rich topsoil (many soil feeders). Epigeal mound structure can differ widely within genera and also between regions within widely distributed species. Macrotermitinae and *Odontotermes* are known to build huge mounds of selected clay-rich subsoil (Wood and Johnson 1986).

2.4.4.4 Arboreal Nesters

Nests are attached outwardly to trees at different heights. These nests are normally made of wood carton. In most cases, the nests are connected to the ground by covered runways. This may assist in distinguishing some arboreal termite nests from those of ants. Nonetheless, some arboreal nesting Nasutitermitinae (e.g., *Hospitalitermes*) form open foraging columns without any connecting runways between the nest and foraging sites (Wood and Johnson 1986).

2.4.5 Termite Communication

Semiochemicals in termite colonies are employed in many activities, such as foraging, defense, attraction to and induction of feeding on food sources, colony segregation, mate location, and even caste regulation (Igwe and Eze 2015). Termites use pheromones, vibrations, and physical contact to communicate with each other. Termites produce many different types of chemical pheromones. Some are volatile and become airborne. Others are not as volatile and remain on surfaces where other colony mates touch them. So their message is transmitted by contact. The alarm pheromone is a volatile type. Contact pheromones eventually dissipate, so each termite that successfully follows the trail lays down another layer of pheromone as it returns to the colony. If the termite did not locate food, moisture, or building materials, no new pheromones are deposited, so the trail eventually fades, and the termites stop following it. Generally, soldiers initiate the trail search for food. By applying a chemical secretion from an abdominal gland (sternal glands located on third, fourth, and fifth abdominal segments) to the ground, they create a short-lived chemical trail leading back to the colony, which the workers follow. They further increase the chemical marking of the trail, resulting in the recruitment of high numbers of nest mates to exploit food sources jointly.

Trail pheromone in termites is not just a single compound but a mixture of compounds. GC/MS and FT-IR analysis of the pheromone extract of *Cubitermes* termites, *Amitermes dentatus*, revealed the presence of fourteen compounds of hydrocarbon (69.22%), ester (14.94%), alcohol (9.15%), and aromatic compounds

(6.70%), the highest being 2-methylundecane followed by 2,7-dimethyloctane (Igwe and Eze 2015). However, the major component of the pheromone appears to be the norsesquiterpene alcohol (E)-2,6,10-trimethyl-5,9-undecadien-1-ol in *Mastotermes darwiniensis*, *Porotermes adamsoni*, and *Stolotermes victoriensis*; the quantity of pheromone required to elicit response is 20 pg/individual in *M. darwiniensis*, 700 pg/individual in *P. adamsoni*, and 4 pg/individual in *Stolotermes victoriensis* with the activity threshold being 1 ng/cm in *M. darwiniensis* and 10 pg/cm in *P. adamsoni*(Sillam-Dussès et al. 2007).

2.5 Life History

The termites are hemimetabolous insects which undergo an incomplete metamorphosis during the process of their growth and development. Unlike most of other insects, very little is known about molting in termites, particularly in the subterranean group. The major reasons attributed to the lack of information include the absence of synchronization in the timing of molt, the cryptic nature of subterranean termites, and their long life cycle. Generally, it takes about 4–5 years for a termite colony to attain its maximum size, with population of more than 60,000–2,00,000 workers.

Termites are opportunistic, small delicate social insects, where they exhibit a trait most aptly by a variety of developmental options available to them. The ultimate caste, or life form, of an immature termite is not determined at the egg stage, and each newly hatched termite can develop into any one of a number of castes. A worker can undergo numerous molts and remain as worker for its entire life span (1–4 years) without changing into another caste. Alternatively, a worker can molt twice, where first it becomes a pre-soldier and then subsequently a soldier. The soldier caste can no longer molt and is considered a final stage. The soldiers have enlarged heads with black jaws. Both workers and soldiers are devoid of wings and usually lack eyes (Myles 2005).

Worker termites can also develop into winged, adult termites (alates), by passing through an intermediate stage called the nymphal stage. Nymphs are termites with external wing buds and are produced on a seasonal basis, usually during the late summer season. As nymphs progress toward the adult stage (i.e., alate), they molt several times to form fully developed wings, eyes, and functional reproductive organs. Nymphs can ultimately molt into sexually mature, fully winged adult male or female alates. Nymphal termites are also capable of a unique developmental pathway wherein they sometimes undergo a regressive molt and lose their wing buds, thereby reverting back to the worker stage. These workers are called pseudergates. A colony may contain several types of neotenic reproductive, wherein secondary (second form) reproductives that develop from nymphs are characterized by the presence of small wing buds and the tertiary (third form) reproductives, which develops from workers, do not possess wing buds (Suiter et al. 2016).

The life cycle of termites commences with the dispersal of winged reproductives to settle and colonize on new resources. The mating pair sheds their wings and thus establishes a new nest. Subsequently, the queen starts oviposition, the egg hatches, and the nymphs develop into different forms (Fig. 2.3). The different castes are determined during the postembryonic development, and each first or second instar termite larva can become a worker, soldier, or reproductive (Laine and Wright 2003). The reproductive caste can have multiple forms (Thorne 1996). The primary reproductives are alate and are designated as the king and queen in the colony. A neotenic reproductive is a reproductive that is not derived from an alate and still retains some juvenile characteristics. Secondary, or nymphoid, reproductives are neotenics derived from nymphs. They have wing pads, and their body pigmentation is generally of a yellow hue. Tertiary or ergatoid reproductives are those derived from false workers (pseudergates). These are generally smaller than nymphoid reproductives and are apterous with very light pigmentation.

2.6 Economic Importance

Termites which inhabit the tropical and subtropical region of the world cause extensive damage to agricultural and horticultural crops, forest trees, stored timbers, wood works in the building, stored products containing cellulose, etc. (Rashmi and Sundararaj 2013). Howse (1970) observed that termites are capable of damaging man-made fabrics (textile materials), plastics (polythene, polyvinyl chloride), and some metal foils.

Among all the known species of termites occurring worldwide, 300 species are considered to be pests (Kumari and Patil 2013), and about 35 species have been reported damaging both agricultural crops and building infrastructure in India (Sharma et al. 2009) causing loss to the tune of US$35.12 million every year (Joshi et al. 2005). However, the annual losses caused by termites in the United States and Japan are US$1000 and US$800 M, respectively (Table 2.3). Harris

Table 2.3 Estimates of annual economic losses caused by termites worldwide

Sl. no.	Country	Annual economic loss
1	Australia	$1.5 billion
2	China (mainland)	$1 billion
3	Fiji Islands	$one million
4	France	$0.5 billion
5	India	$35.12 million
6	Indonesia	$1 billion
7	Japan	$0.8–1 billion
8	Malaysia	$10–12 million
9	Philippines	$100 million
10	Taiwan China	$four million
11	Thailand	$0.5 billion
12	United States	$11 billion
13	**World**	**$40 billion**

(Courtesy: Ahmad et al. 2019)

(1961) reported that the Building Research Institute, Accra, suggested the annual cost of repairing building damage by termite in West Africa is 10 percent of their capital cost and, according to USDA (2010) in Southern United States, the cost of repairing is approximately $1.5 billion each year in wood structure damage. Termites also promote soil erosion by removing the plant cover, thus exposing the soil surface to erosive forces (Harris 1970; Cowie et al. 1989). Harris (1971) documented the damages caused by termites to both earth dams and irrigation ditches.

Roonwal and Chhotani (1967) reported that 58 species of termites cause major damage to wood, whereas sixty-four species of wood-destroying termites were reported by Sen-Sarma et al. (1975), among them 11 being major wood-destroying termites. Seventy-two wood-destroying termite species from Southeast Asia have been reported by Roonwal (1979). About 270 termite species were identified as injurious to economic plants in South Asia (Srivastava 1996). Globally, the estimated loss due to termite damage is about US$50 billion annually (Subektia et al. 2015), although estimates vary considerably according to the cropping systems followed in different geographical regions.

Apart from the negative impacts as discussed above, beneficial aspects of the termites also have to be looked into. They play a key role in decomposition of wood and plant materials and recycling of mineral nutrients to the soil; also, the tunnels they construct in soil are made porously and capable of holding water and organic matter and thus support plant growth. Termites themselves have been used as food in certain parts of the world. In countries where termites are found in large numbers, the inhabitants capture the flying forms for food. The alate termites are nutritious, with a good store of fats and protein (Engel and Kumar 2004). Termites also serve as ecological indicators, where termite knolls have been used as an indicator of underground water; also, a well-developed, active, permanent colony of mound-building termites has been considered as an indicator of underground springs in proximity.

In semiarid and subhumid tropics, significant yield losses are recorded on annual and perennial crops. Damage is more severe during the dry spells in rain-fed conditions than irrigated situation, wherein the plants experience a moisture stress, which predisposes them to termite attack. Among the crops, indigenous one is more capable of resisting the termite incidence than exotic species.

2.7 Nature of Damage

Termites are dreaded polyphagous insect pests that cause damage to wooden structures, forest plants, and agricultural crops. They can attack plants at any stage of development, from the seed to the mature plant.

2.7.1 Termite Damage to Structures

Termites are well known for their ability to damage structures, wood materials, books, and other stored materials of human interest and thus earned the position of pest status for a long time. Magnitude of loss due to termite damage varies according to the type of structural units affected and the species of termite associated with it. The major types of structural damages caused by termites include damage to residential buildings, public places, dams/dykes, protected cultivation structures, and timber in service.

2.7.1.1 Termite Damage in Buildings

Termites construct colony on or in the vicinity of building/structures, and subsequently, their damaging activity can take toll on the integrity of buildings and storage structures resulting into permanent economic losses (Ghaly and Edwards 2011). Arboreal termites belong to genera *Coptotermes* and *Reticulitermes* establish colony in the upper part of a building without need of soil contact; thereby, upper floors of building and timber in service are more prone to damage, whereas the subterranean termites that belong to the genera *Heterotermes and Reticulitermes* were reported to cause massive damage to the buildings by affecting main structure and walls leading to more economic losses up to the abandonment of the affected building (Mahapatro and Chatterjee 2017). Conducive conditions include wood-soil contact (i.e., storing firewood on the ground next to the house, landscape timbers), moisture problems (i.e., leaky pipes, condensation, damaged windows, flat roof, faulty skylights), vegetation along outside perimeter (i.e., ivy, high mulch), dirt-filled porches, improper grade, poor drainage (i.e., downspouts, gutters, spigots), high humidity in crawl spaces, and structural damage due to wind or rain (Green 2018).

2.7.1.2 Termite Damage in Public Places

Termite infestation was reported in many common places, viz., temples, banks, libraries, schools, and museums, from various parts of India. Dry-wood and subterranean termites were reported attacking various archives, libraries, and museums in mostly tropical countries, where books and furniture were included as chief items of display (Pinniger 2012). Dry-wood termites including genera *Cryptotermes* and *Kalotermes* were found to make tunnels in wooden structures and were also reported to inhabit in the stacks of papers and books. Detection could be done by tracing fecal matters in spread condition. Subterranean termites including *Reticulitermes*, *Coptotermes*, and *Macrotermes* are found to attack such constructions through soil and trees.

2.7.1.3 Termite Attack to Dams/Dykes

The structural damages caused by termites to the dams and dykes have been reported from various parts of the world. Tuyen (2006) reported sixty-four species of termites under four families were found from dam sites of Vietnam, majority of which are included under Termitidae. River dams are also found to be attacked by termites in India. The Hirakud Dam on river Mahanadi in Orissa is reported to be infested by termites (Hoon 1960). However, a minor termite infestation by *Odontotermes* species is observed at Hirakud Dam areas (Odisha) in 2012 (Mahapatro and Chatterjee 2017).

2.7.1.4 Termite Damage to Protected Cultivation Structures

The protected cultivation structures for agri-horticultural purposes are often found to be attacked by subterranean termites. Usually, common species reported to cause economic damage in greenhouse, polyhouse, and glasshouse are the Macrotermitinae, under higher termite group. Management of such termites in protected cultivation should be done on contingent basis on both pre- and post-construction phases. Since nylon was found to be more protective against termites, it was recommended for use instead of coir/plastic ones in protected cultivation system. Ropes made of coconut and hemp are recommended to be treated with coal tar creosote by dipping followed by drying, which provide protection against termite attack (Mahapatro et al. 2014).

2.7.1.5 Termite Damage in Timber in Service

Termites were found to attack wood, mostly in dead and decayed stage, but healthy woods were also seen to be susceptible to some termite species. Economic importance of wood-destroying termites involves destruction of timber used in various constructions, their repair, and replacement. Roonwal (1979) listed 83 wood-destroying termites from South Asia, whereas 92 wood-destroying species are recorded (Shanbhag and Sundararaj 2013) from various parts of India.

2.7.2 Termite Damage in Crop Plants

Agricultural crops on which the termite feeds include cereals (maize, sorghum, rice, barley, millet, and wheat), legumes (beans, cowpea, pigeon pea, and chickpea), oil crops (groundnut, sunflower, soybean, and sesame), vegetables (tomato, okra, pepper, and eggplant), root crops (potato, yam, and cassava), fruit plants (guava, coffee, citrus, banana, mango, papaya, grapes, mulberry, pineapple, almond, litchi, and plum), and also sugarcane, cotton, tobacco, and tea (Qasim et al. 2015). Termites cause damage right from the sowing of the crop to harvest. It may also lead to poor germination in crop like sugarcane, wheat, gram, maize, cotton, groundnut, and chilies. However, their incidence in grown-up plants and the yield is reduced drastically because of the losses inflicted at or near maturity, which cannot be compensated by the plants at that stage (Verma and Kashyap 1980). Termite attacks plants in several ways; for instance, the primary and secondary termites attack on *Eucalyptus* species as evident from the tunnels and earthen runways build on the tree surface (Nair and Varma 1981). However, *Odontotermes* species feeds directly on the roots and kills the plant (Harris 1971). Root damage caused by termite interferes with the intake of water, minerals, and nutrients, leading to wilting and drying of the plants. In some cases, *Odontotermes* species infest entire parts of the plant particularly in shrubs and small plants and form earthen sheets and runways on the bark.

The termite worker and soldiers continue their activity under the earthen layer, which covered over the dead barks of the plants (Roonwal 1979). Termite activity can also affect crop quality. The scarification of crop tubers by termites can reduce their market value and increase the toxin content in groundnuts (Jouquet et al. 2018).

The termites are capable of damaging all growth stages of perennial crops and cause significant yield losses (Chhotani 1997). Usually, the attack of termites begins on the roots and then extends to upper parts of the plants. The bark and underlying tissue in older plants are eaten up, which are then gradually exposed to attack of pathogenic microbes, and ultimately result in death of the plant. The bark-eating termites cover the tree trunks and stem surface with a sheet of mud or sometimes make mud galleries on a wide range of crops, rendering them as locally important pests.

Predisposing Factors

According to Paul et al. (2018), the predisposing factors which favor termite attack in crops are as follow as:

(i) The crops grown under unsuitable cropping site and climatic conditions would be stressed and weakened and are more liable to be attacked by termites.
(ii) The accumulation of crop refuge, viz., stubbles, straw, and uprooted dry weeds, in the vicinity of plants serves as additional food resources of termites.
(iii) Non-removal of damaged bark would allow the termites to colonize the pruned dead ends.
(iv) Root damage, due to intercultural operations in field/horticultural crops, emanates root exudates, which serve as attractants for termites.
(v) Root infection caused by soil-borne diseases/nematodes weakens the plants and attracts termites.
(vi) Any stress caused by dry spell in rain-fed conditions, poorly drained soil, etc. favors termite attack.

2.7.2.1 Termite Damage in Field Crops

In field crops, such as cotton and groundnut, the termite eats up the taproots of young seedlings just below the soil surface (Table 2.4). The central root portion is filled with soil, and the damaged plants wilt overnight and die within a few days. In crops viz., sorghum, maize and bajra, termite attacks at the collar region often lead to being toppled down, and when the plant lodges and touches the ground surface, it is exposed to the attack of soil fungi, such as *Aspergillus* spp.

2.7.2.1.1 Termite Damage in Wheat

Wheat is one of the most susceptible cultivated cereal crops to termite attack at all stage of its growth, throughout the rain-fed and irrigated regions in India. Among the termite species, *Odontotermes wallonensis* attacks all stages of wheat plants (Hussain 1935), and the average annual losses are estimated to vary from 6 to 40 percent at different wheat-growing places in India. In field condition, the termite (*Odontotermes obesus*) is the predominant insect pest causing 20–40 percent

Table 2.4 Termite fauna associated with various agricultural crops in India

Sl. no.	Crop	Termite species	States
1	Rice	*Anacanthotermes viarum* (Koenig)	Tamil Nadu
		Microtermes sp.	Delhi and Maharashtra
2	Wheat	*Odontotermes bangalorensis* (Holm. and Holm.)	Madhya Pradesh
		Odontotermes gurudaspurensis (Holm. and Holm.)	Rajasthan and N.W. India
		Odontotermes obesus (Rambur)	Bihar, Delhi, Punjab, U.P., Haryana, M.P., Rajasthan, Gujarat, A.P., Maharashtra, Karnataka, Kerala and Tamil Nadu
		Microtermes obesi Holm.	A.P., Bihar, Delhi, Punjab, U.P., M.P., and Rajasthan
		Microtermes mycophagus (Desneux)	Rajasthan
		Microtermes tenuignathus (Holm.)	
		Eremotermes sp.	
		Nasutitermes sp.	
3	Barley	*Odontotermes gurudaspurensis* (Holm. and Holm.)	Haryana
		Odontotermes latigula Snyder	
		Odontotermes latiguloides (Roonwal and Verma)	
		Microtermes mycophagus (Desneux)	
		Microtermes obesi Holm.	
		Odontotermes obesus (Rambur)	Haryana, U.P., and Rajasthan
4	Oats	*Microtermes obesi* Holm.	Bihar
5	Maize	*Odontotermes gurudaspurensis* (Holm. and Holm.)	Rajasthan and N.W. India
		Odontotermes wallonensis (Wasmann)	Karnataka
		Odontotermes obesus (Rambur)	Rajasthan
		Microtermes obesi Holm.	Bihar and Rajasthan

(continued)

Table 2.4 (continued)

Sl. no.	Crop	Termite species	States
6	Bajra	*Microtermes mycophagus* (Desneux)	Delhi, Punjab, and Rajasthan
		Odontotermes obesus (Rambur)	
		Microtermes obesi Holm.	
		Odontotermes guptai Roonwal and Bose	Rajasthan
7	Jowar	*Microtermes obesi* Holm.	Delhi, Punjab, and Rajasthan
		Odontotermes obesus (Rambur)	Delhi, Haryana, Punjab, and Rajasthan
8	Sugarcane	*Coptotermes heimi* (Wasmann)	U.P. and Bihar
		Eremotermes paradoxi Holm.	Bihar, Delhi, Punjab, U.P., M.P., Karnataka, and Tamil Nadu
		Microtermes beesoni Snyder	U.P.
		Odontotermes taprobanes (Walker)	
		Microtermes mycophagus (Desneux)	Assam and T.N.
		Microtermes obesi Holm.	Assam, Delhi, Punjab, U.P., Rajasthan, Karnataka, Bihar, and T.N.
		Odontotermes assmuthi Holm.	U.P., Maharashtra, A.P., Bihar, and T.N
		Odontotermes bangalorensis Holm.	M.P., Karnataka, and W.B.
		Odontotermes obesus (Rambur)	Punjab, W.B. Assam, Bihar, U.P., Rajasthan, Delhi, Karnataka, M.P., Maharashtra, A.P., and T.N.
		Odontotermes obesus (Wasmann)	Karnataka, A.P., Bihar, M.P., Orissa, and T.N.
		Trinervitermes biformis (Wasmann)	Bihar, U.P., M.P., Maharashtra, Orissa, and T. N.
9	Groundnut	*Microtermes mycophagus* (Desneux)	Rajasthan
		Microtermes obesi Holm.	Rajasthan, U.P., and Delhi
		Odontotermes obesus (Rambur)	Punjab, U.P., Kerala, Rajasthan, Delhi, Gujarat, Haryana, Karnataka, M.P., Maharashtra, A.P., and T.N.
		Odontotermes wallonensis (Wasmann)	Karnataka
		Trinervitermes biformis (Wasmann)	U.P. and Maharashtra

(continued)

Table 2.4 (continued)

Sl. no.	Crop	Termite species	States
10	Tea	*Coptotermes ceylonicus* Holm.	S. India and N.E. India
		Microtermes obesi Holm.	
		Odontotermes obesus (Rambur)	

(Paul et al. 2018)

damage to the crop particularly in rain-fed condition (Mishra et al. 2003). Other important species of termites attacking wheat crop are *O. obesus*, *M. obesi*, and *Microcerotermes tenuignathus*. At vegetative stage, the plants wither and dry up, losing their anchorage and getting dislodged, whereas in reproductive stage (earhead stage), the termite attacks resulting in chaffy earheads with little or no grain. The termite species, viz., *Microtermes mycophagus*, *Odontotermes* sp. nr. *redemanni* and *Odontotermes obesus*, *Trinervitermes biformis*, and *Coptotermes hemi* are found damaging the wheat crop in fields (Gadhiya et al. 2012), and the damage varies from 29 to 100 percent (Parween et al. 2016; Kumar et al. 2018).

2.7.2.1.2 Termite Damage in Maize

Among the termite species, *Microtermes* sp. damages maize crop very seriously; *Odontotermes wallonensis* caused severe damage to young maize plants. Generally, the termite attacks the stem of the maize plant at the ground level, covering with earthen sheet up to a 10 cm height from the base and making a hole on the base, which is completely eaten a few cm upward and downward and filled with soil. However, the outer covering of the stem remained intact. The severely damaged plants are lodged on the ground even by the slightest wind. *Microtermes* sp. attacks maturing and mature maize plants, while *Macrotermes* sp. causes damage to seedlings. Maize plants attacked early in the season can compensate damage with new tiller growth. Agarwala (1955) observed a gradual increase in the intensity of termite attacks from November, when rains ceased. In endemic areas of termite damage, maize-legume intercropping system is the most widely recommended; however, legumes should be selected according to their inherent ability to minimize the termite incidence. The estimated yield loss is about 10 to 48 percent due to termite (Joshi et al. 2005; Mahapatro et al. 2015) from seedling-stage to maturity-stage maize crop.

2.7.2.1.3 Termite Damage in Sugarcane

Sugarcane is mainly damaged by termites belonging to five genera, viz., *Coptotermes*, *Macrotermes*, *Odontotermes*, *Microtermes*, and *Eremotermes.* Agarwala (1955) estimated 2.5 percent loss in sugarcane tonnage and 4.47 percent in sugar production. Termite that causes damage in sugarcane occurs at both seedling (setts) and maturing stage. Among the termite species, *Microtermes obesi* is a serious pest of sugarcane (58%) particularly at seedling stage, whereas Pardeshi et al. (2010) observed a prominent attack of O. *obesus in* sugarcane (43%) in both seedling and maturing stages of the plant. Sugarcane cultivated in the shaded regions of the field are more susceptible to termite attack, where shade, high sugar content, and faster growth rate are some of the major factors, which succumbed to attack of wide varieties of termites. The incidence of termite attack is found lower in sandy loam soil, where the high evaporation rates of sandy loam soil pose a desiccation threat to the soft-bodied termites, which probably restrict their distribution in those areas. According to Parween et al. (2016), the termite species *Microtermes mycophagus* and *Coptotermes hemi* showed 100 percent infestation at seedling stages in sugarcane, whereas *Odontotermes redemanni* showed 100 percent infestation at maturity stage. Vinothkumar et al. (2018) observed that the termite damage (%) in sugarcane varied from 30.67% to 51% in different selected varieties during the period from 60 days after planting to harvest and resulted in reduced cane yield in the range from 16 to 32 percent.

2.7.2.1.4 Termite Damage in Groundnut

The termite species, viz., *Microtermes*, *Odontotermes,* and *Amitermes* spp. cause severe damage in groundnut resulting in a yield loss that ranges from 10 to 30 percent in India and semiarid tropical countries of Africa. Harris (1969) listed 17 termite species found damaging groundnuts in moderate to low rainfall areas of Africa and Asia. They are known to attack groundnut in all stages of growth from seedling to maturity stage, even in storage too. Chhotani (1980) observed the infestation of *O. wallenensis* on groundnut, and typical symptoms are covering of stem with earthen sheet up to 5 cm high from the ground surface. Termites also bore into the main stem just close to the ground level and then tunnel down into the taproot or up to the stem. The damage is also noticed on pegs as well as mature pods, occasionally penetrating into their shells. Vasanthi et al. (2016) observed pod damage (75%) and pod scarification (40.33%) in groundnut by termite species *Odontotermes wallonensis* in Tamil Nadu, India.

2.7.2.1.5 Termite Damage in Cotton

These termite species that belong to three genera, viz., *Trinervitermes*, *Microtermes*, and *Odontotermes*, have been found damaging cotton crop in different cotton-growing regions of India. They feed on the roots and make tunnels in them. The damaged plants show symptoms of wilting because the root tissues are replaced with soil by the termites, eventually leading to death of plants. However, the well-developed root systems of older plants help them to survive the attack of termites.

2.7.2.1.6 Termite Damage in Castor

Castor crop is attacked by termite species *Microtermes mycophagus* at both the seedling and the growth stages. In the seedling stage, the attack is more prominent on roots, where the termites nibble the taproot, but the stem remains unaffected. However, in grown-up plants, the termite infestation has been noticed mainly on stem up to 3 feet from ground level (Parihar 1977). Upadhyay et al. (2012) reported that termite infestation in cotton varied from 9.3 percent (planting) to 31.16 percent (6 month after planting). According to Parween et al. (2016), the termite species *Microtermes mycophagus* accounted for 61.8 percent infestation at seedling stages in cotton, whereas *Microtermes obesi* showed 100% infestation at maturity stage.

2.7.2.1.7 Termite Damage in Pulses

The termite species *O. obesus* is found damaging the important pulse crops, viz., moong bean (*Phaseolus radiates* L.), moth bean (*Phaseolus aconitifolius*), and cowpea (*Vigna sinensis*) at various stages of its growth, and the estimated range of losses is 25–30 percent in cowpea, 10–15 percent in moth bean, and 5–17 percent in moong at Jodhpur, Rajasthan, India (Paul et al. 2018). The termite problem is very serious during very early stage of crop growth in pigeon pea that is responsible for gappy stand in the field and accounted for 1.56 percent of insect pest associated with pigeon pea (Nair et al. 2017).

2.7.2.1.8 Termite Damage in Coconut

The coconut seedlings at nurseries and transplanted seedling in early stages succumbed more to the attack of termite species, *Odontotermes wallonensis*, and serious losses are caused when they nibble at the tender growing point of the plant. It also damages coconut plants in nursery by constructing mud galleries. The tender shoots of coconut seedlings have been observed to be cut off by termites, which also cause damages to roots and shoots of seedlings. The attacked plants dry gradually and finally become wilted.

2.7.2.1.9 Horticultural and Tree Crops

Bark-eating termites are mostly found damaging the various trees grown in orchards. Though the damage they cause is often negligible, in severe infestations, hollowed-out cavities are seen on the main trunk and branches, filled with soil, as well as on roots, filled with soil. The termites collect plant materials and transport them to their nests for their fungus gardens. Sometimes the termites gain entry into the trees through pruned dead ends of branches and stumps. However, in initial stages of establishment of orchards, termites cause more damage by killing the saplings and damaging roots and stems of plants. Termites are known to infest tree crops, such as eucalyptus, silver oak, and casuarina, during the early stages of development and accounted for crop loss to the tune of 10–25 percent (Rajagopal 2002).

2.8 Management

Termites are a highly successful group of eusocial animals, as evidenced by their worldwide distribution and evolutionary persistence (Paul and Rueben 2005). The occurrence of some species in diverse microhabitats confers them an added advantage for thriving in dry, arid, and harsh environments. The cryptic nature and social organization behavior in termites make them difficult to locate and control in a particular environment. Once a subterranean colony is established in an area, it soon invades nearby areas while searching for food and gradually spreads to new locations (Khan and Ahmad 2018). Since most termite pests are subterranean in nature, their management primarily relies on soil treatment with termiticides at the site of attack. But the ill effects of chemical termiticides on the environment call for more focused research on eco-friendly approaches that fit in an integrated management of termites. Any attempts to manage termites need careful consideration of their benefits against the loss to the ecosystem and local communities. Since, termites play major role in decomposing of dead plants and nutrient recycling to soil, they are indispensable member in the food chain. However, a large number of termite species have an economic importance as pests of both agricultural crops and building infrastructure (structural pests). In order to formulate a specific management strategy against termites, a thorough understanding of their biology and ecology is essential. Moreover, the patchiness of infestation under field conditions makes the management strategies more complicated (Paul et al. 2018).

2.8.1 Termite Management in Agricultural Crops

Over the last few decades, various strategies have been developed to manage the termite menace in both fields and horticultural crops, but none of them have proven to be effective in controlling the established termite colonies. Since, termites are always hidden in their nesting colony, any method to eliminate them needs to reach them, but inaccessibility to their niches makes their management all the more difficult. Termites are also known to ward off exposure to the contaminated/treated area. As a defensive strategy, they usually bury the diseased individuals or carry them out of their nests or sometimes eat away the dead ones. Termites are also known to produce certain antibiotics, which allow only their beneficial fungi to grow in their colonies. Hence, the only effective remedy is to prevent their attack and bring their activities to a manageable level. The various strategies used to check the termite activities are broadly classified into nonchemical and chemical methods.

2.8.1.1 Nonchemical Method

It is an alternative to chemical control of termites (control active infestation). This involves a long-term, eco-friendly approach focusing on preventing the termite attack and includes a range of cultural practices physical/mechanical intervention, biological methods, and the use of botanicals.

2.8.1.1.1 Cultural Practices

It is observed that healthy plants may sometimes be damaged by termites, but stressed and unhealthy plants are more susceptible to termite attack (Sen-Sarma 1986). Hence, good-quality seed, healthy seedlings, and appropriate crop management practices should be adopted to maintain good plant vigor (Logan et al. 1990). The proper and balanced application of fertilizer, well-decomposed farmyard manure, frequent irrigation, and use of recommended agricultural practices increase plant vigor, which ultimately reduces termite attacks (Singh and Brar 1988; Negassa and Sileshi 2018), whereas the misuse or incorrect application of commercially available fertilizers may result in accumulation of salts, runoff, leaching, plant damage, and soil pollution (Paul et al. 2018). Though integrated use of manure and fertilizer improves soil fertility and crop yield, prevents soil degradation, and reduces the invasion of termites, the higher dose of fertilizer without organic input increases termite damage (Negassa and Sileshi 2018).

Removing the debris from crop fields helps to reduce food resources that could be used by termites. Thus, crop residues and debris, items made up of wood, cardboard, or paper, which can serve as food for termites, should be removed from the field and building premises (Ibrahim and Adebote 2012; Brown 1962). In termite-prone areas, a higher seed density is recommended so that even when attacked seedlings are thinned out, some amount of economic losses can be avoided (Wood and Cowie 1988). Similarly, before the onset of monsoon, deep summer plowing is recommended in order to destroy the foraging tunnels and mounds where the workers are present in the subterranean region (Kumar 1991).

In large plantations, crop debris and dead woods must be removed on periodical basis, and pruning operations have to be done carefully with clean cuts to minimize the area of exposure. The pruned areas and wounds should be painted with copper oxychloride to avoid termite attacks and dieback (Harris 1971; Sivapalan et al. 1977). Mulching the tree base with vetiver grass leaf reduces the termite attack. Destruction of termite-infested trees and dead trees before the next rainy season helps to prevent movement of swarmers from infested trees, thus removing considerable amount of termite population in the infested area (Srivastava and Butani 1987). The excessive use of nitrogenous fertilizer in tea plantations encourages the growing soft tissues, which are highly susceptible to attacks by *Glyptotermes dilatatus* (Sivapalan et al. 1977). Hence, proper care should be taken while establishing the orchard by avoiding sandy or red sandy loam for growing trees; the pits taken for planting should be treated with soil insecticides, remove the mud galleries in infested tree trunks, and then swab the area with kerosene oil.

2.8.1.1.1.1 Crop Rotation and Intercropping

Intensive cropping, especially monocropping, for long period reduces the soil fertility and the plant vigor. This predisposes the plants to incidence of termites. Hence, farmers should follow crop rotation with non-preferred crops and also follow a cropping system with a fallow period. This helps the soil to regain its fertility status and also to sustain the subsequent crop health, thereby developing some tolerance toward the termite attacks. It is observed that intercropping in maize field with soybean or groundnut reduces the termite incidence and also increases the activity of predatory ants (Sekamatte et al. 2003).

2.8.1.1.1.2 Clean Cultivation

In the area where termite attack is high, both the crop field and surroundings must be cleaned by removing all plant debris to provide protection against termite attack.

Preplanting tillage operations, clearing of weeds in the field bunds, and cultivation along the field borders destroy the termite reservoirs/mounds, thus reducing their foraging activities and ultimately improving plant health (Mahapatro and Chatterjee 2017).

2.8.1.1.1.3 Irrigation Management

Since the water-stressed or sickly plants are more susceptible to termite attacks, healthy plant growth must be sustained by adopting proper management practices. Frequent irrigation reduces attack of *M. obesi* in field crops, such as maize, wheat, sugarcane, and groundnut (Verma et al. 1980; Kumar and Veeresh 1990). But, the activity of termites is found higher in drip-irrigated than in surface-irrigated plots (Jayanthi et al. 1993). The flooding or overuse of water for irrigation reduces plant growth and yield through waterlogging, nutrient leaching and runoff, increased root diseases and weed pressure, and reduced air exchange between soil and atmosphere (Sane et al. 2016).

2.8.1.1.1.4 Soil Management

The regular intercultural operations and preplanting tillage destroy the tunnels and galleries built by termites. These cultural operations restrict their foraging activities and also reduce damage to crops. In black cotton soil, termite incidence is not a problem due to frequent occurrence of small cracks and crevices that prevent maintenance of runways, galleries, and mounds.

2.8.1.1.2 Physical and Mechanical Methods

The management of termites using physical and mechanical means is the common practice followed worldwide to keep the population at a certain level. It includes the following:

(i) De-queening or removing of the king from the colony may destroy the whole colony (Atsbha and Hintsa 2018).

(ii) Destruction of termite mounds through tilling and killing of the primary and secondary reproductives also gives a temporary solution (Tasida and Gobena 2013), since nests are often located deep inside the ground and are difficult to reach. Moreover, this method is highly labor intensive. Chances for some brood and workers to escape along with soldiers are very high, which would eventually develop into large colonies again, after a given time.

(iii) Heating and smoke by burning of the crop debris and residues at the mounds to suffocate the colony are common practice in India (Verma et al. 2018), but it is very difficult to penetrate the smoke and heat deep inside the colony enough to kill reproductive alates as termites have a complex and very sophisticated mound structure. Also, intensive heating makes the soil hydrophobic.

(v) Covering a layer of sand over the nursery area rather than with soil helps to protect coconut nurseries from foraging by the workers (Kashyap et al. 1984). Similarly, digging of deep trenches around the nurseries helps to protect the tree saplings from the incidence of foraging workers of *Macrotermes* sp., as they cannot construct galleries (Beeson 1941). Physical or mechanical barriers are the obstacles used to protect structures from termites. These barriers may be toxic or nontoxic. The physical barrier types, viz., uniform-sized particles and stainless steel screening, are employed as nonchemical methods for managing subterranean termites in the recent years. It was originally discovered by Ebeling and Pence (1957). The barriers composed of soil particles are too large for termites to displace with their mandibles and are too small for termites to pass between them and thus stop termite penetration. The application of gravel sands acts as an efficient physical barrier against two termite species, *Coptotermes formosanus* and *Reticulitermes flaviceps*, under both the laboratory and field conditions (Li et al. 2011).

(vi) Magnets can be used to arrest colony growth in termites, and a strong bar magnets are placed in the soil near the termite mounds to prevent them from further extension of their mound. Diba et al. (2013) used electromagnetic waves to control subterranean termites, *Coptotermes curvignathus* and *C. formosanus*.

2.8.1.1.3 Biological Control

Termites are eaten away by a wide range of vertebrate and non-vertebrate predators, whose natural influence on termite numbers and population dynamics is partially understood in small number of cases (Wood and Johnson 1986). However, natural role of pathogens and other nonsymbiotic organisms is largely obscure (Grasse 1986). Biodiversity of natural enemies can be increased by applying less persistent insecticides and planting large trees around the agricultural fields. Efforts should be made to increase the presence of natural enemies preying on swarms of termites.

2.8.1.1.3.1 Predators

According to Wood and Sands (1978), there exist two different types of predation on termites, viz., on swarming alates and on foraging workers. In the first category of predation type, the antagonists are arthropod predators (which include scorpions, spiders, centipedes, dragonflies, cockroaches, mantids, crickets, beetles, flies, ants, and wasps) and vertebrate predators (including reptiles, amphibians, birds and mammals, sloth bear, echidna, anteaters). In the second category of predator type, foraging termite workers are preyed by ants, such as *Myrmicaria cumenoides* and *Pheidole megacephala (*Sheppe 1970).

Birds (drongo, bush lark, swallow, green bee-eater, hoopoe, Indian roller, etc.) and frogs are important predator of termites during swarming. Reptiles, such as lizards, commonly feed on termites. Mammals, such as the five-striped squirrel *Funambulus pennantii* and mongoose *Herpestes edwardsi*, feed on the swarm of *Microterme* sp. Predatory ants, like *Pheidole* sp. and *Dorylus* sp., are observed feeding on *O. obesus*.

Predatory ants attacking termites belong to families Ponerinae and Myrmicinae (Beeson 1941). Some of the predatory ant species recorded in India are *Leptogenys processionalis*, *Camponotus sericeus*, *Anoplolepis longipes*, and *Oecophylla smaragdina* (Rajagopal 1979; Kumar 1991), and the ant species, such as *Iridomyrmex purpureus* (Oberst et al. 2017), *Plagiolepis pallescens*, *Polyrhachis* *lacteipenni*, *Pheidole teneriffana*, *Crematogaster antaris*, and *Monomorium destructor* (Latifian et al. 2018), are found preying on termites in Chinese agricultural ecosystems.

2.8.1.1.3.2 Pathogens

Microbial control is the exploitation of microorganisms (such as bacteria, fungi, nematodes, and viruses) and their by-products against insect pests. However, the biological control of termites with microbes gained much attention during the past decades. Many researchers evaluated the effectiveness of entomopathogenic bacteria, fungi, and nematodes against termites. The entomopathogenic fungus (*Metarhizium anisopliae*) is promising (Abonyo et al. 2016), and it can be used as a potential biopesticide against termites. Some bacterial species, such as *Bacillus* spp., provided promising results against termites (Kalha et al. 2014). Similarly, some species of entomopathogenic nematode (*S. riobrave*, *S. carpocapsae*, *S. feltiae*, and *H. bacteriophora*) are also found causing mortality in subterranean termites (Yu et al. 2010). Minimal work has been done for the use of viruses in biological control; however, nuclear polyhedrosis virus (NPV) can be exploited as an important microbial agent against termites (Zhang and Mo 2014).

2.8.1.1.3.2.1 Bacteria

Among the entomopathogenic bacteria belong to Bacillaceae, a considerable work has been done on evaluating the efficacy of *Bacillus* spp. (*B. thuringiensis* and *B. thuringiensis* subsp. *israelensis*) against termites and found that they are very promising in managing the termites (*Microtermes obesi* and *Microcerotermes beesoni*) (Singha et al. 2010). Fifteen species of bacteria have been tested against the termite *Coptotermes formosanus*, and among them, *Serratia marcescens* Bizio is found effective in both field and laboratory conditions (Osbrink et al. 2001). Both *Bacillus subtilis* (Omoya and Kelly 2014) and *B. licheniformis* (Natsir and Dali 2014) showed good pathogenicity against termites when used as feeding baits and sprays. Some species of rhizobacteria produce hydrogen cyanide (HCN) that can be used for the management of subterranean termites (Natsir and Dali 2014). The HCN-producing rhizobacteria (i.e., *Aeromonas caviae*, *Alcaligenes latus*, *Rhizobium radiobacter, Pseudomonas fluorescens*) are found effective under in vitro conditions against *Odontotermes obesus* (Devi 2012; Devi and Kothamasi 2009).

2.8.1.1.3.2.1 Fungi

Among the entomopathogenic fungi, those that belong to genera *Antennopsis*, *Beauveria*, *Coreomyceptosis*, *Laboulbenia*, *Leboulbeniopsis*, *Metarhizium*, *Metirolella*, *Neotermus*, and *Termitaria* are pathogenic to termites. However, their pathogenicity relies on ability of their asexual spores or conidia to disperse, transmit, adhere, germinate, and penetrate the host cuticle for fungal growth and infection and compete with other fast-growing opponents and cause disease outbreak in the termite colony (Yii et al. 2015). The fungal spores may be applied as a powder or through baits, either on termites or on nests. Extensive studies have been carried out worldwide to evaluate *Beauveria bassiana* and *Metarhizium anisopliae* against termites. The fungi *M. anisopliae* is found effective in controlling different termite species such as *Coptotermes formosanus*, *Odontotermes* sp., and *Reticulitermes* sp. (Dong et al. 2009). The isolates of *M. anisopliae* showed the pathogenicity toward termite, *Odontotermes* sp. (Balachander et al. 2009), and the application of *M. anisopliae* spores along with the attractants, such as sugarcane bagasse, sawdust, and cardboard, brings about 95 percent mortality in workers and more than 75 percent in soldiers within 13 days (Balachander et al. 2013). Ravindran et al. (2015) evaluated the efficacy of four isolates of *M. anisopliae* against *Coptotermes formosanus*, and these are found effective in bringing mortality after 4 days.

Sileshi et al. (2013) tested four isolates of *M. anisopliae* and *B. bassiana* against *Macrotermes* sp. and observed 100 percent mortality after 7 days and suggested that they can be used as environmentally safe biopesticides. The fungal spores (*M. anisopliae*, *B. bassiana*, and *Paecilomyces fumosoroseus*) applied along with farmyard manure (FYM) at the time of sowing provided better germination, survival, and yield in maize plants (Rana and Dinesh 2014). Application of *Aspergillus* sp. and *Isaria fumosorosea* caused rapid mortality in termites (Pandey et al. 2013; Wright and La 2013).

Although many researchers have confirmed that entomopathogenic fungi are promising biocontrol agents in managing various termite species, few studies proved their failure for termite management. Since termites are highly sensitive to environmental abiotic factors, they have very well-developed multi-defense strategies against fungal pathogens, and they prevent the entry of pathogens into their colonies by avoiding fungus-infected areas and fungus-infected individuals, releasing antifungal secretions and using symbiotic microorganism for nest materials (Liu et al. 2019). While in case of pathogens entering into the colony, a well-developed olfactory sense plays a key role in identifying and removing lethal conidial spores (Yanagawa et al. 2009). Then, they eliminate pathogens quickly by grooming (Poulsen 2015), antifungal secretions, aggressive behavior, cannibalism, burial (Liu et al. 2019), and social immunization (Liu et al. 2015; Cremer et al. 2018). Further, detoxification also plays an important role in improving social immunity (Cremer et al. 2018; Liu et al. 2019).

2.8.1.1.3.2.1 Virus

Although some viruses (NPV and GV) have great potential to be good microbial control pathogens against agricultural insect pests, they have gained very little attention worldwide (Chouvenc et al. 2011). Little attempts have been carried out to investigate the efficacy of viruses against termites under laboratory conditions. NPV is thought to be an important microbial agent against termites. However, its effectiveness against termites under field conditions is yet to be explored. Zhang and Mo (2014) determined the toxic effects of *Autographa californica* NPV (AcNPV) on workers of *Coptotermes formosanus*. They treated filter paper or *Pinus massoniana* wood dust with different concentrations of NPV and fed these to termites. Their results revealed that AcNPV has a strong infection capacity against termites. Further, they suggested that *P. massoniana* wood dust treated with NPV is a very effective practice for termite control.

2.8.1.1.3.2.1 Nematodes

Entomopathogenic nematodes (EPNs) are very important parasites and have the potential for controlling many agricultural insect pests residing in soil habitat. These pathogens are used as biological control agents to kill Macrotermitinae (Lin et al. 2015). Though they are very effective against termites under laboratory conditions, none of them are found useful under field conditions. The EPNs invade different body structures of termites, such as nervous and muscle tissue, fat body, and salivary and sternal glands. The nematode species, such as *Heterorhabditis baujardi* and *Heterorhabditis indica,* are found parasitic on termites under both field and laboratory conditions at Egypt (EL-Bassiouny and El-Rahman 2011). The families of EPNs, such as Steinernematidae and Heterorhabditidae, are the important obligate insect parasites and can be used as efficient pathogens against termites. Yu et al. (2010) used three strains of *Steinernema riobrave* against subterranean termite species *C. formosanus*, *Heterotermes aureus*, and *R. flavipes* and observed that *S. riobrave* is more effective against *Heterotermes aureus* after 4 days of treatment. Shahina and Tabassum (2010) conducted laboratory evaluation studies to reveal the efficacy of *Sphingobacterium pakistanense* against subterranean termite *Macrotermes* and found it effective in controlling the termite, whereas Murugan and Vasugi (2011) used a mixture of EPNs and neem seed kernel extract against *R. flavipes* and obtained more effective results (Rathour et al. 2014).

2.8.1.1.4 Botanicals

Botanicals are plant-based products or plant-derived compounds that are considered as the most potential substitutes for highly harmful synthetic pesticides, and it includes the compounds present in plant roots, stems, leaves, flowers, fruits, seeds, and wood. More than 2000 species of plants belonging to 60 families have pesticidal activities; many of them are used as insect growth regulators, ecdysteroids, behavior modulators, feeding deterrents, repellents, attractants, and so on (Verma et al. 2018).

Many researchers have investigated the effect of plant-derived materials against termites under both laboratory and field conditions. The leaf extract of four plant species *Heliotropium bacciferum*, *Lantana camara*, *Rhazya stricta*, and *Ruta chalepensis* is found effective against subterranean termite *Psammotermes hybostoma* (Alshehry et al. 2014). Similarly, the plant extract of both *Maesa lanceolata* and *Azadirachta indica* is effective in controlling termites infesting the hot pepper (Ibrahim and Demisse 2013). Seo et al. (2014) estimated the fumigant toxicity of four plants, viz., *Chamaemelum nobile*, *Eriocephalus punctulatus*, *Ormenis multicaulis*, and *Santolina chamaecyparissus* against the termite *Reticulitermes speratus*. Among the plants, *Chamaemelum nobile* resulted comparatively higher mortality in termites after 2 days of treatment. The leaf extract of *Azadirachta indica* and *Jatropha curcas* is a potential biopesticide against termites against *Macrotermes* spp. (Addisu et al. 2013). The methanolic extract of *Jatropha curcas* (Verma et al. 2013) and both bark and leaf extracts of *Lawsonia inermis* (Adedeji et al. 2017) is found effective in reducing the termite attack on various trees. The root extract of *Crotalaria burhia* and the leaf extract *Anacardium occidentale* are effective in repelling the termite species, *O. obesus*, infesting different plants (Bajya et al. 2015).

The seed extracts of *Chenopodium ambrosioides*, *Maesa lanceolate*, and *Vernonia hymenolepis* (Addisu et al. 2013) and leaf extract of *Camellia oleifera*

(Hu et al. 2015) are effective against *Macrotermes* sp. and *Reticulitermes flavipes*, respectively. Similarly, the leaf extracts of *Lantana camara* have antifeedant, repellent, and toxic effects against *Reticulitermes flavipes* (Yuan and Hu 2012). The monoterpenes from aromatic plants (Xie et al. 2014) and neem oil also are found inducing mortality in different termite species. The essential oils from four plants, *Chamaemelum nobile*, *Eriocephalus punctulatus*, *Ormenis multicaulis*, and *Santolina chamaecyparissus*, have potential fumigant toxicity on termites (Himmi et al. 2013), and it is effective against the Japanese termite *Reticulitermes* (Seo et al. 2014). The essential oils derived from marigold and sweet orange have strong repellency against termites (Verma et al. 2016). Similarly, the essential oils from seven plant species viz. *Corymbia citriodora*, *Croton sonderianus*, *Cymbopogon martini*, *Lippie alba*, *L. gracilis*, *L. sidoides*, and *Pogostemon cablin* are effective against worker cast of *Nasutitermes corniger* (Lima et al. 2013a, 2013b). The novel carbohydrate-based enzyme inhibiters cellobioimidazole (CBI) and fluoromethylcellobiose (FMCB) are found potential against *R. flavipes* (Zhou et al. 2008). Some plants are resistant to attack of many subterranean species of termites (Pandey et al. 2012). Two plant species *Khaya ivorensis* and *K. senegalen* exhibit natural resistance against termites (Franca et al. 2016).

Numerous significant works have been done on phyto-based substances against insect pests during the past decades that obtained satisfactory results, but the compatibility with chemical insecticides, formulating them to commercial available form, and their efficacy under field conditions are still need to be exploited.

2.8.1.2 Chemical Methods

2.8.1.2.1 Insect Growth Regulators

Two classes of insect growth regulators (IGRs), juvenoids (juvenile hormone analogues or JHAs and juvenile hormone mimics or JHMs) and chitin synthesis inhibitors (CSIs), have been tested on termites. The gradual and cumulative mode of IGRs' action makes them promising candidates for incorporation in baits. Unlike most insect species in which juvenile hormones (JH) are responsible for retaining the immature forms, the effect of JH in termites is unique. Luscher (1958) for the first time suggested that JH regulated soldier formation in termites.

The application of JHAs induces significant soldier formation in termite species that consists of lower natural soldier proportions (e.g., 1%–2% for *Reticulitermes* spp.) than for species with higher proportions (e.g., 10%–20% for *Coptotermes* spp.) (Su and Scheffrahan 1990). However, the field trials using JHAs, such as methoprene and hydroprene, have failed to yield an increase in soldiers or a colony decline in *Prorhinotermes simplex* (6.9%–22.2% soldier proportion; Haverty 1977) (Hrdy et al. 1979), whereas the application of another JHA, fenoxycarb, against field colonies of *Reticulitermes* species increases the number of pre-soldiers and soldiers initially and subsequently declines their foraging activity (Jones 1989).

Derivatives of benzoylphenylureas are known to inhibit the chitin synthesis of insects, other arthropods, and fungi (Hajjar and Casida 1978). Doppelreiter and Korioth (1981) demonstrated the ecdysis inhibition by difubenzuron (Dimilin) against *Heterotermes indicola* (Wasmann) and *Reticulitermes flavipes* for the first time. However, subsequent testing with diflubenzuron on field colonies of *Microcerotermes* species provided inconclusive results (Faragalla et al. 1985). Laboratory studies have indicated that CSIs such as diflubenzuron (Su and Scheffrahn 1993) or lufenuron (Su and Scheffrahn 1996) inhibited the ecdysis of *R. flavipes* but caused virtually no effect on *C. formosanus*. The CSI hexaflumuron is known to cause significant ecdysis inhibition of a wide range of economically important subterranean termites species, including *Reticulitermes*, *Coptotermes*, and *Heterotermes* species (Su and Scheffrahn 1993, 1996).

2.8.1.2.2 Synthetic Insecticides

Even though chemical insecticides have ill effects on the environment, farmers across the world use them extensively for managing the pest problems in agricultural crops. Management of termites with termiticides is a difficult task as termites are eusocial insects and live in mounds or many inches below the soil surface to keep them protected from outside threats. However, some termiticides, such as imidacloprid, chlorpyrifos, fipronil, spinosad, chlorfenapyr, bifenthrin, cypermethrin, permethrin, and chlorantraniliprole, have been used worldwide for the management of termites. Farmers use these termiticides during irrigation of the crops; however, injection of termiticides is also recommended to control termites in field crops, forests, and buildings. Some fumigants, such as carbon dioxide, methyl bromide, sulfur fluoride, and phosphine, are also applied through fumigation methods for controlling the termites attacking dry woods and stored grain products. Among the six insecticides (imidacloprid, indoxacarb, fipronil, spinosad, thiamethoxam, and chlorfenapyr) evaluated against *Microtermes mycophagus*, both chlorfenapyr and spinosad are the most potent and bring about mortality in due course of time when compared to other insecticides (Iqbal and Saeed 2013).

The insecticides, viz., chlorpyrifos, acetamiprid, and thiamethoxam are found effective in controlling the termite *Psammotermes hypostoma* (Ahmed et al. 2015), whereas clothianidin efficiently reduces termite infestation on sugarcane sets, which is statistically on par with the combined application of acephate+imidacloprid (Bhagawati et al. 2014), whereas imidacloprid, a non-repellent insecticide, causes more than 90 percent mortality in *Microtermes obesi* at laboratory condition and reduces the tunnel numbers and cumulative tunnel distance at open fields (Manzoor et al. 2014). Fipronil and spinosad are found effective in managing the subterranean termite *Heterotermes indicola* (Saljoqi et al. 2014). The insecticides abamectin, ivermectin, fipronil, and imidacloprid showed higher toxicity against termites *Reticulitermes speratus* under laboratory conditions (Chen et al. 2015). Lufenuron induces sublethal effects on *Coptotermes formosanus* physiology and behavior and significantly reduces survivorship, running speed, food consumption, and tunneling and also inhibits the carcass-burying and particle transport behavior of the termites (Wang et al. 2014). Both spinosad and spinetoram exhibit toxicity and non-repellency against termites, *Coptotermes formosanus*, under laboratory conditions (Bhatta et al. 2016). Cyclohexylamine is a novel termiticide, and its hydrogen phosphate salts maintain its toxicity against termites (Grimball et al.

2017). Though termites can be effectively controlled through onsite application of chemical insecticides, we cannot ignore their injurious effects on humans, animals, and birds and the fact that indiscriminate use of pesticides leads to pest resistance and environmental pollution.

2.8.1.2.3 Method of Application of Chemical Insecticides and Insect Growth Regulators

2.8.1.2.3.1 Soil Barrier

This method involves creating a chemical barrier in the soil, which is toxic to termites on contact, and it also has repellent characteristics. The effectiveness of the termiticides over a long period of time is achieved when applied as a continuous barrier to and under the foundation especially to manage subterranean termites. If there are untreated gaps in the soil, termites may circumvent the chemical treatment. Termiticides that act by creating a chemical barrier in the soil include bifenthrin, cypermethrin, and permethrin.

2.8.1.2.3.2 Treated-Zone Termiticides

They are non-repellent but show delayed toxicity as termite forage through the treated soil, which they do not avoid. The termites contact the active ingredient as they penetrate the "treated zone," which causes delayed mortality and also allows the termites to be overcome by lethal microbes. The toxicant is passed to the nest mates through grooming activities and social food exchange (trophallaxis). The control is usually achieved within 3 months. It includes termiticides, viz., fipronil, imidacloprid, and chlorfenapyr.

2.8.1.2.3.3 Dusting

Dusts can be used as standalone treatments or as a part of a combined approach. The dust settles on the termites, which then carry back to the nest. The termites clean themselves of the dust and ingest the active ingredient. The active ingredients are spread throughout the colony, through grooming which is a social activity by the termite. If enough active ingredient is ingested, then it is possible to eliminate the entire colony. The active ingredients in the dust may include arsenic trioxide, fipronil, and triflumuron. Scheffrahn et al. (2001) reported that newly established colony of dry-wood termites, *Cryptotermes brevis*, could be prevented using dust formulations of commercially available disodium tetrahydrate.

2.8.1.2.3.4 Fumigation

Fumigants (e.g., sulfuryl fluoride) treat all infestations simultaneously and have high level of efficacy, if correctly applied. The fumigant, sulfuryl fluoride, kills the dry-wood termites in about 3 days. The advantage of fumigants over localized treatment is that it may eliminate infestations hidden from view and necessitates to vacate the structure within 2–3 days of treatment.

2.8.1.2.3.5 Baits

Bait technology uses wood or cellulose matrix favored by termites that is impregnated with a slow-acting toxic chemical as active ingredient. Termite workers feed upon the bait and transfer it by grooming activities and social food exchange (trophallaxis) to other colony members, eventually reducing or eliminating the entire colony. Bait system can be applied as standalone treatments or as part of a combined approach. Bait products that are available commercially include Sentricon® termite colony elimination system (hexaflumuron), Firstline® termite defense system (sulfluramid), Subterfuge® termite bait (hydramethylnon), and Outpost® termite bait response (diflubenzuron).

2.8.2 Management of Termites Damaging Various Structures

Since termites are one of the most important pests damaging the residential buildings, nonresidential structures, and other materials on which human beings are greatly dependent, it is necessary to develop its eco-friendly management strategies which are always safe to man and other beneficial organisms. In India, wood is an inseparable constituent of constructions ranging from buildings for dwelling or other purposes, worship places, agricultural constructions, and social and facilitation constructions which are highly vulnerable to the attack of termites. According to Rawat (2013), the popular management practices, which are focusing on preventing the termite attack in various structures, include pre-treatment of the construction site, pre-treatment of construction materials, using physical barriers, chemical control, baiting techniques, using biological control method, and many more processes. The various approaches to manage structural termite pests are given below.

2.8.2.1 Barrier Treatment

Both physical and chemical barrier treatments can be provided to prevent the attack of termites to various structures.

2.8.2.1.1 Physical Barrier

The fine material particles, viz., sand and crushed volcanic cinders, can be used as physical barrier against subterranean termite, *Reticulitermes hesperus*. This barrier is impenetrable for termites because of heaviness of the particles for termites to carry away and acts as disruptors for building gallery (Logan et al. 1990). It is recommended to install the transparent plastic sheet just below damp proof course level of new buildings, which can protect the construction from termite attack and dampness. Crushed stones, gravel aggregate, and sand are also used as physical barriers against subterranean termites, and it should be applied in the crawling space, viz., under the pillars, beam foundation, and terrace which serve as good physical obstacle for termites attacking buildings (Rawat 2013). Since humidity plays a major role in influencing the termite survival, installing dehumidifier in entry and crawling space of termites in a construction followed in the United States found effective in giving protection to the building from termite attack (Anonymous 2015).

2.8.2.1.2 Chemical Barrier

Chemical barriers are classified into two categories, viz., repellant and non-repellant. Non-repellant chemicals used against termites include imidacloprid, chlorfenapyr, chlorantraniliprole, and fipronil, whereas chemicals with repellent action include bifenthrin, chlorpyrifos, and permethrin. While using repellant chemicals, termites can detect them and move away from the treated area, resulting in non-transfer of toxicity to the other colony members, whereas in case of non-repellants, toxicity cannot be detected earlier resulting in passing the lethal effect to the other colony members (UNEP, 2014). As per recommendation of Bureau of Indian Standards (2013), soil treatment with chlorpyrifos 20% EC at the time of building construction acts as a chemical barrier against termite's attack. Another popular practice of termite management in buildings of India is reticulation or tubing system in which network of pipes is laid beneath the floor. According to Joshi (2002), the pipes that contain termiticide act as chemical barrier to which chemicals are pumped inside with drippers and it can be used as and when needed. "Perimeter-treatment" can also be followed by digging shallow trench/channel and application of termiticide solution at 1.75 l per running meter and for buildings with concrete or masonry apron; drilling 12 mm holes at 30 cm distance close to plinth wall and injection of termiticide solution 2.25 l/hole are suggested to protect from termite attack (Mahapatro and Chatterjee 2017).

2.8.2.2 Baiting Techniques

Baiting is a promising technology of managing termites damaging various structures. Here, chitin synthesis inhibitors are used as active ingredient, and the same is applied through food matrix or as dust. Baiting technique is almost proved against lower termites including *Reticulitermes* and *Coptotermes*, and it is proved successful in managing *Reticulitermes santonensis* in Paris (UNEP 2014). Toxicants used in baiting techniques are divided into three groups, viz., (i) the metabolic inhibitors, (ii) biological control agents, and (iii) insect growth regulators (IGRs) (Ngee et al. 2004). Slow-acting poison baits are helpful in reducing large number of termite individuals as the lethal chemicals get sufficient time to be spread to the larger number of individuals in a colony. Accumulation of dead termites due to quick effective baits may act as the mark of danger (=alarm) to other nest mates, resulting in avoidance of bait station (Rawat 2013).

2.8.2.3 Borate Technique

Borate refers to the compounds containing both boron and oxygen, which naturally exists in the form of sodium and calcium borate. In North-East Indian state Sikkim, bamboos are widely used as building material, and treatments with borate confer protection to bamboo against termite attacks. Forest Research Institute (Dehradun), India, has recommended the use of borate compounds for wood preservation in the form of chomated copper borate (CCB) and ammoniacal copper zinc borate

(ACZB). Since application of boron to soil leads to leaching problems, boron treatment is recommended for the structures above ground level (Mahapatro and Chatterjee 2017).

2.8.2.4 Botanical Control

Various phytochemicals and secondary metabolites which possess toxic or repellent effects on termites, viz., alkaloid, terpenoids, and unsaturated isobutylamide, can be used for pre-treatments of wooden house and building materials, such as wooden frames, pillars, logs, and timbers. The extracts of *Cannabis sativa*, *Datura alba*, *Curcuma amada*, *Ricinus communis*, and *Asafoetida* sp. are reported as effective against termites. The application of anti-termite herbals and local application of those in termite-affected sites yielded good results in India, but their application and persistence in soil treatment against subterranean termite attack to buildings need further research (Verma et al. 2009). Though botanicals are comparatively less effective than chemicals, they are biodegradable, nontoxic, and environment friendly in nature. In the ancient Vadakkumnathan Temple of Thrissur, Kerala, the wooden crafts and other timber in service damaged by termite could be successfully managed by using a mixture of locally available phyto-based products and oils (total of eight ingredients), and the temple was honored with UNESCO Asia–Pacific Awards for Cultural Heritage Conservation for the year of 2015 (Prakash 2015).

2.9 Conclusion

Termites are the most dominant organism in tropical ecosystems. This is mainly due to their ability of utilizing dead plant materials rich in cellulose (the most abundant organic matter on the earth). With this, they become important in the processes, viz., decomposition of organic matter, vital link in many food chains, soil engineering (translocating and altering soils physically and chemically; maintaining soil fertility), providing possible input for nitrogen fixation, methane gas release, and carbon flux. In some arid and semiarid areas, activities of termites result in fertile areas, in an otherwise harsh environment. The potential and ability of termites to conquer harsh environments and their resilience during high levels of disturbance are as impressive as the sophistication and beauty of structures they create.

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Aphids

3

Rajendra Singh and Garima Singh

Contents

Abstract

The aphids are soft-bodied small insects (< 7 mm) and feed by sucking plant sap. They usually live in colonies on the undersides of leaves or tender terminal shoots. Aphids excrete considerable amount of sugary liquid, honeydew, on which sooty mould usually turns them black and also serves as food for ants, bees and their parasitic wasps. The aphids are unique on the account of their peculiar mode of reproduction, development and polymorphism. They may reproduce either by parthenogenesis, zygogenesis or paedogenesis. They may either be oviparous or viviparous and alatae or apterae, the male often being wanting and frequently rare in certain generations. Parthenogenetic reproduction allows rapid increase in numbers and results in populations consisting of clones. Some species reproduce both parthenogenetically and sexually (holocyclic species), whereas only few reproduce parthenogenetically (anholocyclic species). In parthenogenetic reproduction, life cycle completes within 10 days in temperate

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R. Singh (✉)
Department of Zoology, Deen Dayal Upadhyay Gorakhpur University, Gorakhpur, India

G. Singh
Department of Zoology, University of Rajasthan, Jaipur, India

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regions. The aphids are polymorphic, and both winged (alate) and wingless (aptera) morphs may be found in the same colony. Several factors, both biotic and abiotic, have effect on the formation of different phenotypes. Each morph performs different ecological roles in the life history which is characteristic of aphids. This trait coupled with the ability to breed by means of diploid parthenogenesis and viviparity for a major part of the life cycle in aphids has enabled them to produce a large number of clones in different kinds of plants even under adverse conditions. Aphids are frequently engaged in mutualistic associations with bacterial endosymbionts that not only provide essential amino acids to them but also grant them protection from natural enemies, protection from extreme temperatures, development of resistance to a fungal pathogen and the ability to use a greater diversity of resources. Out of globally 5110 species of aphids described, about 250 species are major agricultural and horticultural pests. They damage the crops directly by sucking their nutrients, making galls and hampering photosynthesis and respiration by the growth of sooty moulds on the honeydew deposited thereon. Aphids also damage the crop indirectly by transmitting hundreds of plant viruses. Because of their economic importance, their population must be controlled to save the crops. In this contribution, several aspects of aphid systematic and biology such as endemism, host–plant association, diversity, morphology, feeding behaviour, life history, polymorphism and factors affecting it, migration, defence, aphid–ant association, endosymbiosis, economic importance and their population management have been described in detail.

Keywords

Aphids · Systematics · Life cycle · Polymorphism · Aphid-plant interaction · Economic importance · Control practices

3.1 Introduction

Aphid, also known as greenfly, blackfly, plant lice, ant cow, etc., is a common name for all the bugs that belong to the family Aphididae (order Hemiptera, suborder Sternorrhyncha, infraorder Aphidomorpha, superfamily Aphidoidea). They are small (1–10 mm), soft-bodied, sap-sucking insects infesting both aerial and subaerial parts of a variety of plant species, few of which are notorious pests of agricultural and horticultural crops. They possess a proboscis which originates between and behind the forelegs; their antennae have two thick basal segments and a flagellum composed of up to four segments, the last of which is divided into a proximal part and a thinner distal part called a ‘process terminalis’. They possess two compound eyes and two ocular tubercles made up of three lenses each which are situated behind and above the compound eyes. They have two tarsal segments; the wings when present have only one prominent longitudinal vein. The fifth abdominal segment bears a pair of upward and backward pointing tubes on the dorsal surface called siphunculi or cornicles, and a cauda is usually present below and between them on the last abdominal segment. They are cosmopolitan but are most abundant in

Fig. 3.1 A parthenogenetic aphid colony consisting of individuals of different morphs (alate and apterous) and viriginoparae giving birth along with attending ants

temperate climates. They are unique on account of their peculiar mode of reproduction, development and polymorphism. They may reproduce either by parthenogenesis, zygogenesis or paedogenesis. They may either be oviparous or viviparous. The sexes may be unequally represented (males are frequently rare) in certain generations. Parthenogenetic reproduction allows rapid increase in numbers and results in populations consisting of clones. Some species reproduce both parthenogenetically and sexually (holocyclic species), whereas only a few reproduce solely through parthenogenesis (anholocyclic species) (Dixon 1977, 1998; Singh and Ghosh 2002) (Fig. 3.1).

Aphids are related to members of other Aphidomorpha, e.g. adelgids (conifer aphids, pine aphids, spruce aphids) (family Adelgidae, superfamily Adelgoidea) and phylloxerids (family Phylloxeridae, superfamily Phylloxeroidea) (Heie and Wegierek 2009), all of which probably evolved about 280 million years ago (MYA) in the Carboniferous; they probably bred on nonflowering plants such as Cordaitales and Cycadophyta (Capinera 2008). The oldest known fossil aphid is *Triassoaphis cubitus* from the Triassic about 220 MYA (Evans 1956). Aphids did not always look like they do now; the characteristic shape and wing venation and the structure of their proboscis had evolved by the Jurassic (e.g. *Juraphis crassipes*), whereas the cauda and the siphunculi evolved later in the Cretaceous about 55 MYA (Shaposhnikov 1979).

3.2 Systematics and Biology

There have been different opinions regarding the nomenclature of the aphid taxonomy. The classification of aphids is still not well established that may be accepted all over the world. There were various opinions regarding the classification of the Aphidoidea. Aphid taxonomy is often frustrated by the host alternation and extensive polyphenism displayed by many species. In the literature, some aphidologists (Remaudiere and Remaudiere 1997; Blackman and Eastop 2007) refer to the families of Heie (1987) as subfamilies. There is no extremely good reason to prefer one categorisation over the other, and fortunately this dichotomy in the literature has led to little confusion. However, the use of family designations has the only single advantage of allowing slightly more details in taxonomic hierarchies. Favret and Eades (2020) in aphid.speciesfile.org kept all the extant Aphididae under 24 subfamilies, viz. Aiceoninae, Anoeciinae, Aphidinae, Baltichaitophorinae, Calaphidinae, Chaitophorinae, Drepanosiphinae, Greenideinae, Eriosomatinae, Hormaphidinae, Israelaphidinae, Lachninae, Lizeriinae, Macropodaphidinae, Mindarinae, Neophyllaphidinae, Phloeomyzinae, Phyllaphidinae, Pterastheniinae, Saltusaphidinae, Spicaphidinae, Taiwanaphidinae, Tamaliinae and Thelaxinae. Table 3.1 summarises the species diversity of aphids in different taxa of Aphididae in the world and in India.

3.2.1 Endemism in India

Globally, 5110 species of aphids are described under 528 genera. Out of 24 subfamilies of Aphididae, only 16 subfamilies are represented in India (Table 3.1). In India, 809 species of aphids under 208 genera are reported, out of which about half of them (at least 385 species) are endemic, and among them almost all are represented in the Himalayas region. The most species diversity was observed in the subfamily Aphidinae (444 species) (Singh et al. 2014, 2015a; Singh and Singh 2016a, b, c, 2017a, b, c) followed by Greenideinae (96 species) (Singh and Singh 2017d), Eriosomatinae (64 species) (Singh and Singh 2017e) and Hormaphidinae (57 species) (Singh and Singh 2018). Singh and Singh (2019) summarised the diversity of Indian aphids. At least 32 endemic genera are represented in India. Except *Aspidophorodon* Verma, *Indiaphis* Basu, *Neomasonaphis* Ghosh and Raychaudhuri, *Myzakkia* Basu and *Brachyunguis* Das, all other endemic genera are monotypic. All the eight species under Aiceoninae, three species of Taiwanaphidinae and one species of Lizeriinae are endemic. The percentage of endemism in other subfamilies is as follows: Thelaxinae (75%), Greenideinae (71.9%), Calaphidinae (62.5%), Drepanosiphinae (50%), Hormaphidinae (47.4%), Aphidinae (44.5%), Chaitophorinae (44.4), Lachninae (39.0%) and Eriosomatinae (34.4%). Only eight endemic species are present in the peninsular area including two species, viz. *Eutrichosiphum davidi* and *Paoliella nirmalae*, which are exclusive of this area. The high percentage of endemism shows that the Himalayan areas provide congenial ecological conditions for the active speciation of aphids, while the

Table 3.1 Subfamily-wise distribution of extant aphid species recorded in India and abroad

Subfamilies		World		India		Endemic species	References
		genera	species	genera	species		
Aiceoninae		1	18	1	8	8	1
Anoeciinae		2	30	1	6	2	1
Aphidinae	Aphidini	33	832	9	69	14	2–5
	Macrosiphini	244	2264	96	375	178	5–11
Baltichaitophorinae		1	1	0	0	0	–
Calaphidinae	Calaphidini	17	77	5	14	11	12
	Panaphidini	46	271	20	34	19	12
Chaitophorinae	Chaitophorini	7	153	4	27	12	1
	Siphini	5	25	1	2	0	1
Drepanosiphinae		5	39	2	4	2	1
Eriosomatinae	Eriosomatini	14	113	5	21	9	13
	Fordini	19	83	7	15	6	13
	Pemphigini	21	178	7	28	7	13
Greenideinae	Cervaphidini	6	20	3	7	2	14
	Greenideini	7	152	5	87	64	14
	Schoutedeniini	3	7	1	2	1	14
Hormaphidinae	Cerataphidini	10	113	9	34	13	15
	Hormaphidini	5	12	2	2	0	15
	Nipponaphidini	29	105	11	21	14	15
Israelaphidinae		1	4	0	0	0	–
Lachninae	Eulachnini	4	294	2	21	8	16
	Lachnini	4	33	4	10	4	16
	Stomaphidini	1	33	1	1	0	16
	Tramini	3	31	2	2	1	16
	Tuberlachnini	5	15	3	7	3	16
Lizeriinae		5	41	1	1	1	16
Macropodaphidinae		1	7	0	0	0	–
Mindarinae		1	9	1	2	0	16
Neophyllaphidinae		1	18	0	0	0	–
Phloeomyzinae		1	1	0	0	0	–
Phyllaphidinae		4	18	1	1	0	16
Pterastheniinae		2	5	0	0	0	–
Saltusaphidinae		12	57	1	1	0	16
Spicaphidinae		2	13	0	0	0	–
Taiwanaphidinae		1	14	1	3	3	16
Tamaliinae		1	6	0	0	0	–
Thelaxinae		4	18	2	4	3	16
Total		**528**	**5110**	**208**	**809**	**385**	

1. Singh and Singh 2016d; 2. Singh and Singh 2016a; 3. Singh and Singh 2016b; 4. Singh and Singh 2016c; 5. Favret and Eades 2020; 6. Singh and Singh 2017a; 7. Singh and Singh 2017b; 8. Singh and Singh 2017c; 9. Singh et al. 2014; 10. Singh et al. 2015a; 11. Singh et al. 2015b; 12. Singh and Singh 2017f, 13. Singh and Singh 2017e; 14. Singh and Singh 2017d; 15. Singh and Singh 2018; 16. Singh et al. 2018

peninsular region which is a part of the Gondwanaland is very old and stable landmass with distinct flora and fauna quite different from the northern parts.

3.2.2 Host–Plant Association in India

Singh and Singh (2016a, b, c, d, 2017a, b, c, d, e, f, 2018; Singh et al. 2014, 2018, 2015a) recently catalogued the Indian aphids and their food plants. Of all plant species, 25 % are used as food plant by the aphids, and though it is believed that the speciation of aphids has followed that of plants, not all groups of plants are equally infested (Mahr 2018). The Asteraceae, the third most specious plant family, supports the most aphid species (696 species) (Singh et al. 2015b), but the Orchidaceae, the second most specious plant family, supports only nine species of aphids, while the Rosaceae which is only the 22nd most specious plant family supports the third highest number of aphid species (293 species) in India. The plant family which supports the second highest number of aphids is Coniferae (includes several families, viz. Pinaceae, Araucariaceae, Podocarpaceae, Sciadopityaceae, Cupressaceae, Cephalotaxaceae, Taxaceae [363 species]), but these are nonflowering plants. Most aphids are monophagous, though some species are highly polyphagous, e.g. *Myzus persicae* which feed on more than 500 different plant species worldwide; in India, it alone infests 293 plant species under 64 plant families (Singh et al. 2015a).

Few aphids are known to make gall on the plants. The aphid galls are abnormal outgrowths of the plant tissues that serve as their own microhabitats as well as food. Galls may also provide the aphid with physical protection from parasitoids and predators (Fig. 3.2). In Northwest Himalayas, about 76 gall-inducing aphid species

Fig. 3.2 A gall on a leaf made by gall making aphid (**a**) and a gall cut to show the aphids inside (**b**). Courtesy bugguide.net

are reported under Eriosomatinae, Hormaphidinae, Aphidinae and Calaphidinae in these areas, and all of them are present in Northwest Himalaya. But Northeast Himalaya supports only eight gall-inducing species (10.52%), and most of them belong to Aphidinae. Of Eriosomatinae, 93% can induce galls in Northwest Himalaya (Chakrabarti 2007). In general, the gall-forming aphid species are heteroecious, i.e. alternate between their primary and secondary hosts in different periods of the year. However, a few have been found to be autoecious, i.e. monophagous. These aphids are also highly polymorphic in nature. Unless morphs from both of their primary and secondary hosts are available, their identities in some cases are difficult (Chakrabarti 1987).

Availability and diversification of host–plants has direct influence on the diversification of aphids. The major host–plant subclasses that harbour more aphid species are Rosidae, Asteridae, Dilleniidae and Colelinidae. More than 150 aphid species are found on these plants. Out of about 696 plant species described under the family Asteraceae in India, only 207 species were found to be infested by 199 aphid species. Among the aphid species, *Aphis gossypii* was recorded feeding 77 species of Asteraceae, followed by *Brachycaudus helichrysi* (72 species), *Aphis spiraecola* (70 species), *Myzus persicae* (45 species), *Myzus ornatus* (35 species), *Aphis fabae* (25 species) and *Aphis craccivora* (23 species). *Artemisia* and *Sonchus* were observed to be attacked by 60 and 25 aphid species, respectively (Singh et al. 2015b).

There are many examples of special host associations in this area. The nonavailability of specific primary host has influenced the life cycle patterns in many species especially under the subfamily Eriosomatinae. Many species continued anholocyclic parthenogenesis for a long time, such as species of Fordini and Hormaphidinae. Recently, Singh and Singh (2019) have summarised the diversity and food plant associations of Indian aphids.

3.2.3 Diversity in India

The great diversity and abundance of aphids usually occur in the higher altitudes where subtropical to warm temperate climate prevails which represents transition area between Oriental and Palaearctic realms. The biological diversity of India is well reflected in the distribution and abundance of fauna and flora and also in aphid–host association. The richness and diversity of aphid fauna is largely due to diverse flora in different kinds of ecosystem present in India. The food plants of Indian aphids cover over 1250 species belonging to 137 plant families and 86 orders and suborders (Agarwala and Ghosh 1985) against a total plant species of about 45,000 found in India. Among the areas enjoying subtropical to warm temperate climate in India, diversity and concentration of aphids are more pronounced in Northwest and Northeast Indian states, whereas hotter and drier areas of Indo-Gangetic and Peninsular India have less species diversity and poor prevalence. Chakrabarti (2009) analysed the aphid diversity, its distribution and endemism, host–plant association and the life cycle pattern from subregion of the Oriental region and observed that all these are different in each zone. Out of the total Indian aphid species, the Himalayas represent 808 species in 219 genera under 17 subfamilies indicating high species richness. The Northwest Himalayas has 573 species in 177 genera of 16 subfamilies, and the Eastern Himalayas has 464 species in 147 genera of 14 subfamilies, while Central Himalayas is represented by only 67 species in 56 genera of 8 subfamilies. The Peninsular India has 126 species in 56 genera, and the Gangetic plain has 64 species in 32 genera, while the Indus valley has only 27 species in 15 genera (Chakrabarti 2009).

Subfamily-wise breakup of the taxa in the said biogeographical areas reveals that in Northeast India, subfamily Anoeciinae represents 8 species, Aphidinae 270 species, Chaitophorinae 7 species, Drepanosiphinae (including Calaphidinae, Lizeriinae, Phyllaphidinae, Saltusaphidinae and Taiwanaphidinae) 24 species, Greenideinae 49 species, Hormaphidinae 29 species, Lachninae 11 species and Eriosomatinae 16 species. Similarly, in Northwest India, subfamily Anoeciinae represents 8 species, Aphidinae 168 species, Chaitophorinae 13 species, Drepanosiphinae (including Calaphidinae, Lizeriinae, Phyllaphidinae, Saltusaphidinae and Taiwanaphidinae) 28 species, Greenideinae 24 species, Hormaphidinae 12 species, Lachninae 23 species and Eriosomatinae 34 species. On the other hand, Gangetic plain represents Aphidinae 42 species, Greenideinae 2 species and Calaphidinae 1 species only. Also, Indus plain represents three subfamilies, viz. Aphidinae 30 species, Calaphidinae 1 species and Greenideinae 1 species. Lastly, in Peninsular India, Aphidinae represents 50 species, Calaphidinae 4 species, Greenideinae 10 species, Hormaphidinae 5 species, Lachninae 1 species and Eriosomatinae 3 species. Thus, among five biogeographical areas, Northeast India represents the maximum number of aphid species and subspecies (414), followed by Northwest India (310), Peninsular India (73), Gangetic plain (45) and Indus plain (32). Also, the subfamily Aphidinae among other subfamilies (Anoeciinae, Drepanosiphinae, Calaphidinae, Lizeriinae, Phyllaphidinae, Saltusaphidinae, Taiwanaphidinae, Greenideinae, Hormaphidinae, Lachninae and Pemphiginae) has higher frequency of occurrence than any other subfamilies. The Peninsular India having Eastern and Western Ghats and Vindhya Range as well as Gangetic and Indus plains has comparatively poor diversity of aphid fauna. It may be mentioned here that both Northeast and Northwest India represent the genera belonging to all subfamilies. The subfamily Anoeciinae is apparently yet to be recorded from Peninsular India. Likewise, the subfamilies Aphidinae, Greenideinae and Calaphinae are on record from both Peninsular and Gangetic plain regions. It is worthwhile to mention that the genera belonging to subfamily Aphidinae are found to occur in all the regions of India and its genera are most abundant in all the biogeographical areas in comparison to other subfamilies of the family Aphididae (Ghosh and Singh 2000).

3.2.4 Morphology

The body of the aphid is usually divisible into the head, thorax and abdomen. However, in some species, it is very difficult to divide the body due to tendency of fusion of the segments. The external morphology of an aphid is provided in Figs. 3.3 and 3.4. The head is usually dorsoventrally flattened. The number of antennal segments varies between one and six. The last antennal segment has a stout base and a short to very long slender terminal portion, the processus terminalis, with at least three terminal hairs. The primary rhinarium is placed at the junction of the base. The eyes are always well developed and larger in the winged morph than wingless ones. At the posterior margin of the eye protrudes an ocular tubercle, or triommatidion. Alate aphids bear three ocelli, one on the front of the head and the other two laterally near the anterior part of each eye. The proboscis, which is laid back beneath the body when not in use, may be so long, especially in the species that live on trees that it sticks out beyond the end of the abdomen. The stylet bundle with which aphids take up plant sap consists of two pairs of needlelike stylets, the inner pair of maxillary stylets and the outer pair of mandibular stylets (Fig. 3.3).

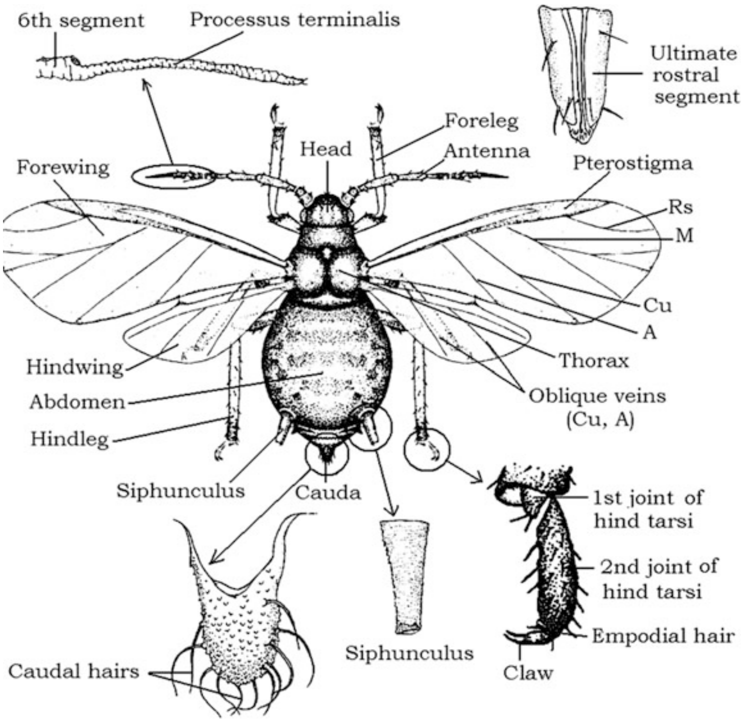


Fig. 3.3 Aphid showing different parts of the body. Wing venation: *A* anal, *Cu* cubital, *M* median, *Rs* radio sector

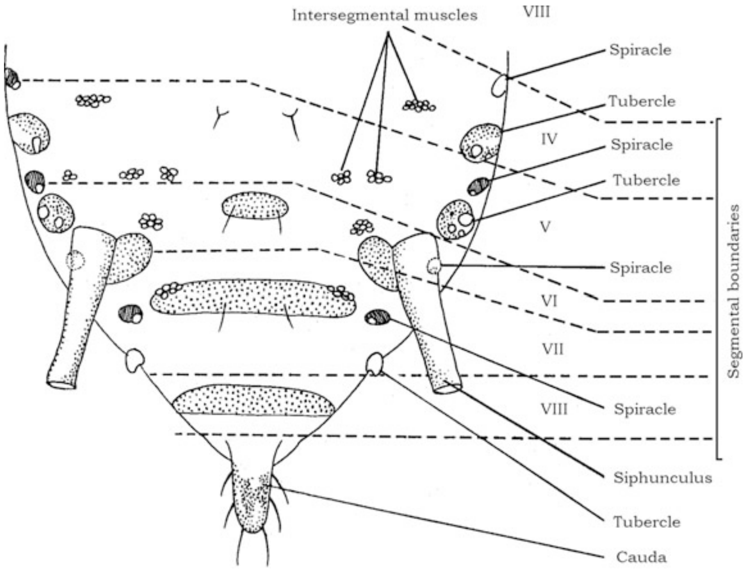


Fig. 3.4 Posterior part of the abdomen of an aphid

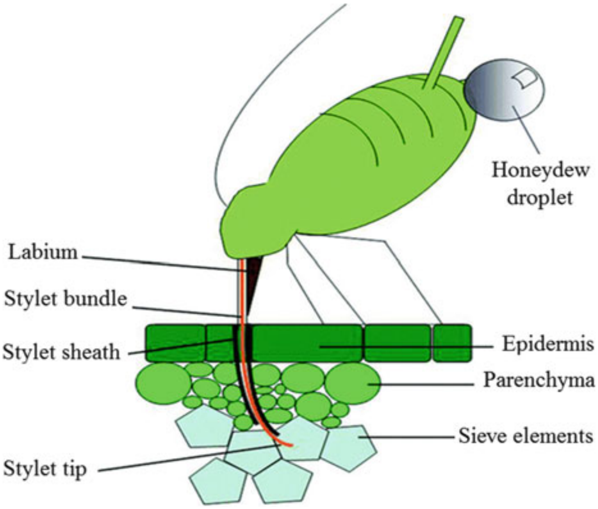
The prothorax or the entire thorax may be fused variably with the head. Each thoracic segment bears a pair of legs having usually five segments: coxa, trochanter, femur, tibia and two-segmented tarsi. The segments are partly or completely fused together or atrophied in some species, especially in Pemphiginae and Hormaphidinae. The alatae bear a pair of wings; these are of similar consistency, but the forewing is always longer and broader than the hindwing. The forewings have two longitudinal veins: one is the costa, which is a weak vein running along the frontal edge of the wing, and the other is the strong main vein which runs just behind the costa (Fig. 3.3).

The abdomen consists of nine visible segments, the ninth being the cauda (Fig. 3.4). The first segment may be fused with the thorax or may remain distinct; other segments except the segment 8 may be clearly or indistinctly demarcated from each other or may get fused. Aphids bear nine pairs of spiracles (or stigma) on their bodies: one pair on the meso- and metathorax and on the first seven abdominal segments laterally. Wax plates or pores may be present on the dorsum of the thorax and abdomen, the position and size being variable in wax plates. Because of this, the aphids are often more or less pulverulent with white waxy exudates, wither of powdery, filamentous, plate-like or rod-like appearance. Small papilla-like tubercles are often observed in Aphididae. They occur singly as a rule on each segment and are arranged on the body in a marginal row and less often also occur in mesal and pleural rows. The ventral hairs may be short to long, and they are usually with cute apices. Dorsolaterally on the abdominal segments 5 or 6 usually occurs a pair of siphunculi or cornicles which are typically tube-like structure through which alarm pheromones exudate (Behura 1996a). These may be altogether absent, but when present they may be of variable shapes and sizes, viz. ring-like, mammiform, cone-shaped, cylindrical, truncate, tapering, slightly to distinctly clavate or cigar-shaped, and may be with a flange near its apex; the siphuncular opening may be placed right at the apex or may be shifted laterally; and the surface may be smooth or warty. At the posterior end of the abdominal sternum, there are two sclerotised plates, the anal plate which represents the tenth abdominal sternite and the genital plate which represents the eighth. Subanal plate is situated ventral to the cauda which may be entire, indented, elongate, oval, semioval, crescent-shaped, knobbed or bilobed, semicircular, broadly triangular, semicircular, etc. The subgenital plate bears many hairs. The cauda is short, either crescent-shaped, semicircular, broadly triangular or shortly tongue-shaped, and bears two to many hairs (Fig. 3.4).

3.2.5 Feeding Behaviour

Both nymph and adult aphids feed in the same way. They are phloem feeder. They find the phloem vessels from the stems, leaves and roots of the plant. In most cases, they feed passively by means of high pressure within the sieve elements of the plant (Fig. 3.5). The maxillae and mandibles are elongated into a stylet bundle that penetrates the plant tissues to reach the feeding site in the phloem. At this time, the distal tip of the labium helps stylet penetration from the outside, acting as a guide. The stylets, enclosed within the proboscis when the aphid is not feeding, are very thin and could break during insertion into the plant. Therefore, aphids secrete a substance from the tips of their stylets which begin to harden forming a hard protective covering around the stylets as they are slowly pushed into the plant in search of the phloem tubes (Miles 1999; Will and Vilcinskasa 2015). The saliva also isolates plant tissues from the mouthparts avoiding plant reaction at the feeding site

Fig. 3.5 Schematic representation of a feeding aphid (Guerrieri and Digilio 2008)



(Felton and Eichenseer 1999). When the stylets reach a phloem tube, the aphid injects saliva into it. The saliva helps prevent the plant cell from sealing the puncture (i.e. the mouthparts of the aphids) with special proteins which are the plants' normal defence mechanism (Will and van Bel 2006; Pettersson et al. 2017). Aphids pierce their stylets very slowly, and it may take half an hour to 24 hours from beginning to prick the stylets to actually sucking the phloem juice. This feeding habit causes little mechanical damage to the plant as compared to that of biting and chewing insects. The phloem sap also contains high sugar level that causes high osmotic pressure inside the stomach of the aphid due to which water transfers from haemolymph to the stomach causing hyperosmotic stress that may cause the death of the aphid. Therefore, the aphids avoid this situation by several osmoregulatory mechanisms (Ashford et al. 2000). Excess sugars are excreted through anus called honeydew which is used by other insects as food.

3.2.6 Life History

The life history of aphids is highly complicated including parthenogenetic and sexual generations, elaborate polyphenism and obligate shifting between unrelated host–plant taxa. These and other unusual life cycle traits occur in a variety of combinations among the approximately 5100 extant species within the family. The aphids have prolific breeding, polyphagy, advanced degree of polymorphism, anholocyclic/holocyclic reproduction, host alternation and high potential for rapid evolutionary changes because of parthenogenesis and polyvoltinism (Minks and Harrewijn 1987; Behura 1994).

Some aphids are anholocyclic in which males are totally absent, and the parthenogenetic diploid females reproduce only by viviparity, while others living in temperate climates are holocyclic that produce males and oviparae, which mate to produce eggs for overwintering. Anholocyclic ones overwinter with viviparous females in protected locations, and no sexual morphs and eggs emerged. According to the aphid evolution theory, anholocycly originated from holocycly during the fourth glacial epoch (Moran 1992). Dixon (1998) and Hardie (2017) illustrated the generalised life cycle pattern of aphids and discussed the role of nutrition on the production of morph. Aphid life cycles are complex and may be either monoecious or dioecious, involving holocycly or anholocycly. In the simple and generalised monoecious holocyclic aphid life cycle (Fig. 3.6a), the aphids feed on a single host–plant species throughout the year. The sexual morphs are produced in the following autumn, in response to decreasing photoperiod. Then, mating takes place between males and females (oviparae) producing genetically recombinant eggs that overwinter on the host–plant. In the spring, fundatrix emerges from the eggs that overwinter on the host–plant and mature parthenogenetically and gives births to nymphs that mature to viviparae and continue the reproduction by this way in the summer. If the aphid group produces plant galls, the fundatrix is responsible for their production. The viviparae may be apterae (wingless) or alate (winged), but in some groups all viviparae are alate. The parthenogenetic reproduction of viviparae allows very rapid

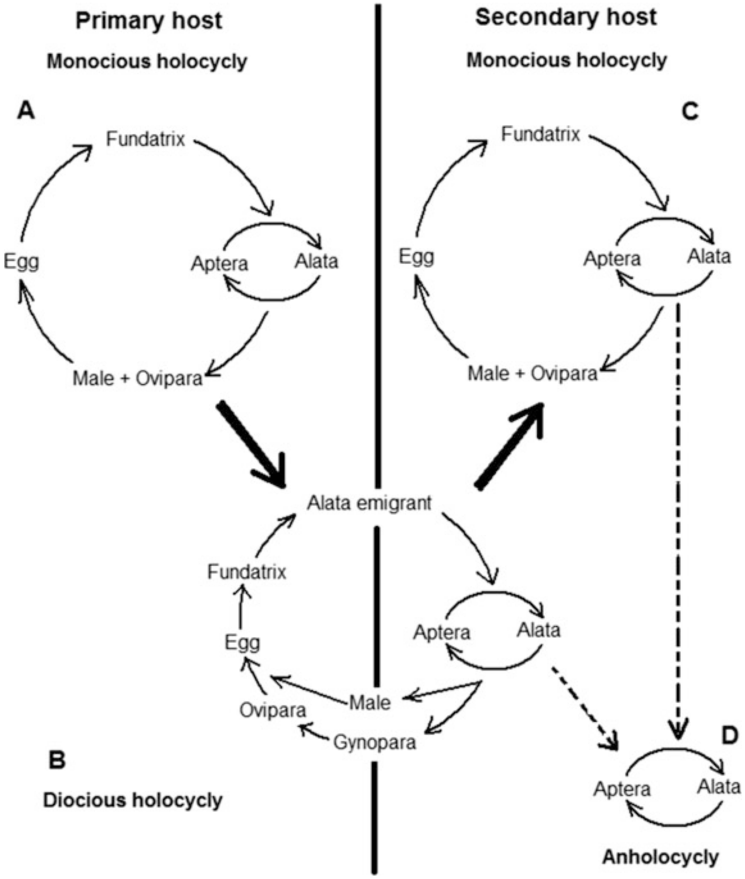


Fig. 3.6 Evolutionary development of generalised aphid life cycles. Initially, aphids developed monoecious holocycly on an ancestral woody primary host, where aestivation occurred because sap amino acids were unavailable during summer growth cessation (**a**). Next, multiple subfamilies independently evolved dioecious holocycly, where viviparae moved to summer-growing herbaceous secondary hosts but returned to their ancestral host in autumn (**b**). In some aphids, secondarily monoecious holocycly developed on the secondary host when the primary host was lost (**c**). Often in warm areas, where selection for an overwintering egg is not imposed, some populations of dioecious and secondarily monoecious holocyclic aphids may lapse into facultative anholocycly on their secondary hosts; this condition may become obligate anholocycly if the ability to produce sexuals is lost (**d**) (Sorensen, 2009)

buildup of numbers and collapse of generation time. Apterae produce more offspring per female than do alatae. Once an aphid population reached its maximum, due to crowding effect among apterae or lack of nutrient levels, they turn to produce alatae, which migrate to better situations. During migration, a number of individuals die off because of landing on unsuitable food plants. In the situation, when alatae find a suitable host–plant, they fed for a short period, and they reproduce viviparidae generation. The production of viviparae continues until autumn when the conditions stimulate them to produce sexuals.

A second, more complicated dioecious life cycle (Fig. 3.6b) has independently evolved among several different aphid groups that show seasonal alternation between differing hosts. This dioecious cycle probably evolved in response to the seasonally inadequate supply of nutrients, especially amino acids, on their primary host. Woody deciduous plants normally transport amino acids in quantity only during the spring. Aphid groups feeding on and confined to such plants face a nitrogen deficit during the summer, when active plant growth retards and phloem sap is low or lacks nitrogen. In this situation, some groups of aphids, e.g. *Periphyllus* spp., may develop an aestivating nymph that halts growth until autumn, while others, e.g. Aphidinae, have developed to escape those primary hosts during the late spring. Their spring alatae migrate to herbaceous fast-growing secondary hosts as emigrants during the summer. In the autumn, when the secondary hosts disappeared, the aphids come back to their woody primary host by producing migrating males and gynoparae. The male and oviparae mate to lay their overwintering eggs. Depending on the aphid or its group, their secondary host–plants are either specific or of several plant species, but the primary hosts are often specific to a plant genus. However, in warmer climates, the aphid populations do not need an egg for overwintering survival. Under such conditions, otherwise holocyclic dioecious or monoecious populations may drop facultatively into anholocycly on their secondary hosts. If such populations remain anholocyclic for a longer period, they evolve into obligate anholocycly by losing the capacity to produce sexual morphs (Dixon 1998).

Some aphid groups have evolved beyond dioecious holocycly, entirely leaving their primary host to remain on their secondary host, in secondarily monoecious holocycly (Fig. 3.6c). These aphid groups do not require eggs to overwinter survival. Under such conditions, otherwise holocyclic dioecious or monoecious populations may lapse facultatively into anholocycly on their secondary hosts (Fig. 3.6d). If such populations remain anholocyclic long enough, they may eventually evolve into obligate anholocycly by losing the ability to produce sexual morphs, despite undergoing environmental conditions that normally trigger their production (Dixon 1998; Hardie 2017).

About 80% of the described species from India are parthenogenetic virginoparous for most of the year but are capable of sexual reproduction with production of eggs (Singh and Ghosh 2012). They develop in parthenogenetic female without fertilisation. Even embryos inside parthenogenetic females may contain embryos, i.e. a mother can have in its ovarioles developing embryos which in turn also contain embryos, the future granddaughters. Thus, there is a telescopic generation due to parthenogenesis and viviparity in aphids (Minks and Harrewijn 1987). This results in reduced postnatal development periods and generation time. All aphids have diploid parthenogenesis, and there is no reduction division, and development starts from germinal cells with full complement of chromosomes including XX chromosome. Sexual females, like asexual ones, have two sex chromosomes, i.e. XX. Males have only one sex chromosome, i.e. OX. In theory, this means males could produce sperm with either no sex chromosomes, i.e. an O, or one sex chromosome, i.e. an

X. However, in reality, sperm with an O sex chromosome degenerate very rapidly and never contribute to an embryo. This means that all offspring of a sexual mating must have XX as their sex chromosomes, because females always contribute an X chromosome, and therefore all aphids resulting from sexual matings are female. Eggs are laid during the autumn as the overwintering stage in many temperate forms and, as explained above, give rise to females whether they are the result of sexual mating or not. In other species, a special overwintering form develops in the autumn called a 'hiemalis', while in some species the adults are the overwintering stage. Ova within a viviparously reproducing female start to develop immediately after ovulation; this occurs long before birth (even human females are born with all the ova they will ever need throughout their life, though they remain undeveloped for many years). This means that an embryo can exist inside another larger and more mature embryo. In fact, a newly born summer aphid can contain within herself not only the developing embryos of her daughters but also those of her granddaughters which are developing within her daughters. Parthenogenesis combined with this 'telescoping of generations' gives aphids an exceedingly rapid turnover of generations, meaning they can build up immense populations very quickly. There is a more or less regular cyclic or anholocyclic alternation of parthenogenetic oviparous and viviparous generations associated with polymorphism, changes of food plants and mode of life. Several generations often succeed each other, in which the males are extremely rare or are totally absent. Individuals of the same generation often differ considerably from one another. Some have fully developed wings, others have atrophied wings and still others are apterous (Singh and Singh 2016).

A generalised life cycle pattern of heteroecious life cycle of bird cherry aphid, also known as oat aphid, *Rhopalosiphum padi*, is illustrated in Fig. 3.7 whose several populations in temperate regions that have cold winters reproduce combining sexual and asexual phases (holocycly), alternating between primary winter tree hosts (bird cherry) and secondary summer host–plants that are grasses (Poaceae). In contrast, the reproduction of populations in areas where the winters are mild, and also in some temperate countries where the primary hosts are less available or absent, the life cycle is completely parthenogenic (anholocyclic) on poaceous plants.

Although low temperature, short day length and physical condition of host–plants are regarded as important factors governing the production of sexuales, the discovery of apterous oviparous females of the mustard aphid *Lipaphis erysimi* on mustard in the arid and semiarid region of Jaipur (Rajasthan) (Ghosh and Rajendran 1988) suggests that some other factors are also operative in the phenomenon. Complete life histories of Indian aphids are not known. The possible life cycle of *Lipaphis erysimi* (Fig. 3.8) in India is illustrated. Medda et al. (1997) studied different modes of life cycles of some aphid species infesting *Salix* in India (Fig. 3.9).

3.2.7 Polymorphism

Aphids are remarkable on account of their peculiar mode of development and the polymorphism, i.e. occurrence of two or more morphologically distinct morphs in a

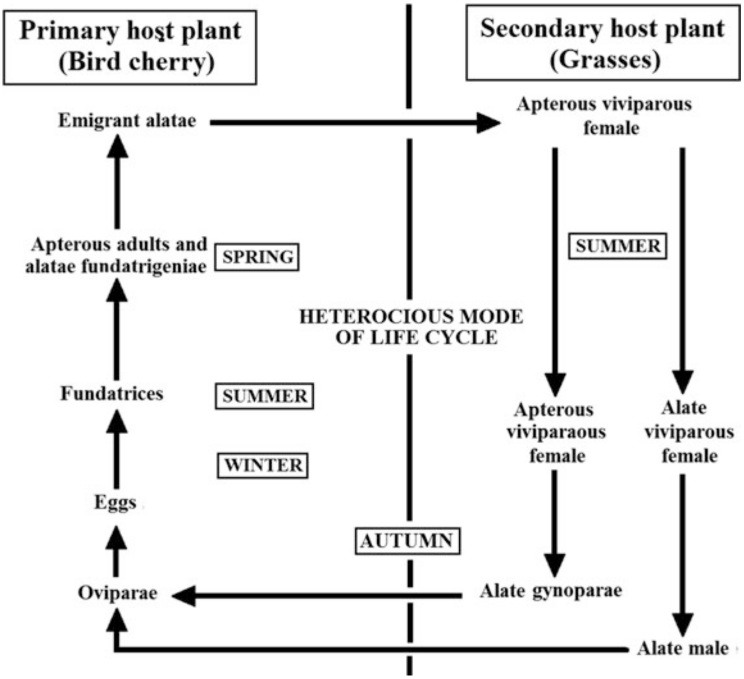


Fig. 3.7 Generalised heteroecious life cycle of oat aphid, *Rhopalosiphum padi*

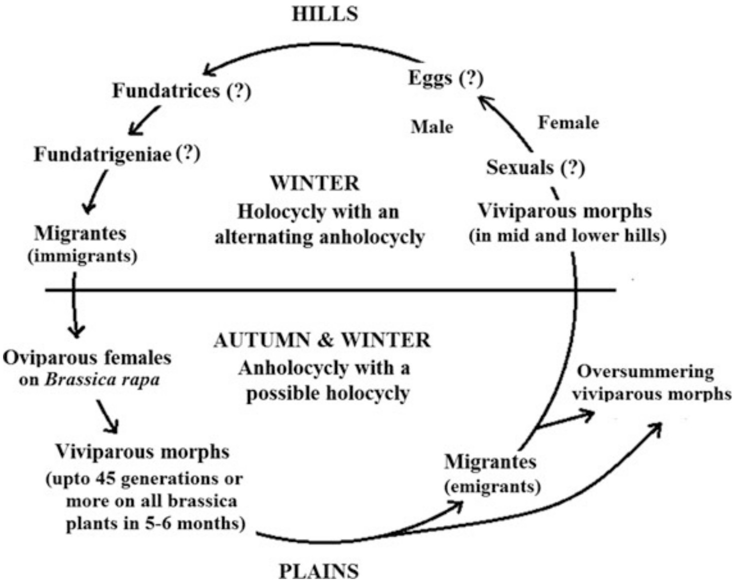


Fig. 3.8 Possible life cycle of the mustard aphid, *Lipaphis erysimi* in India (Singh and Singh 2016)

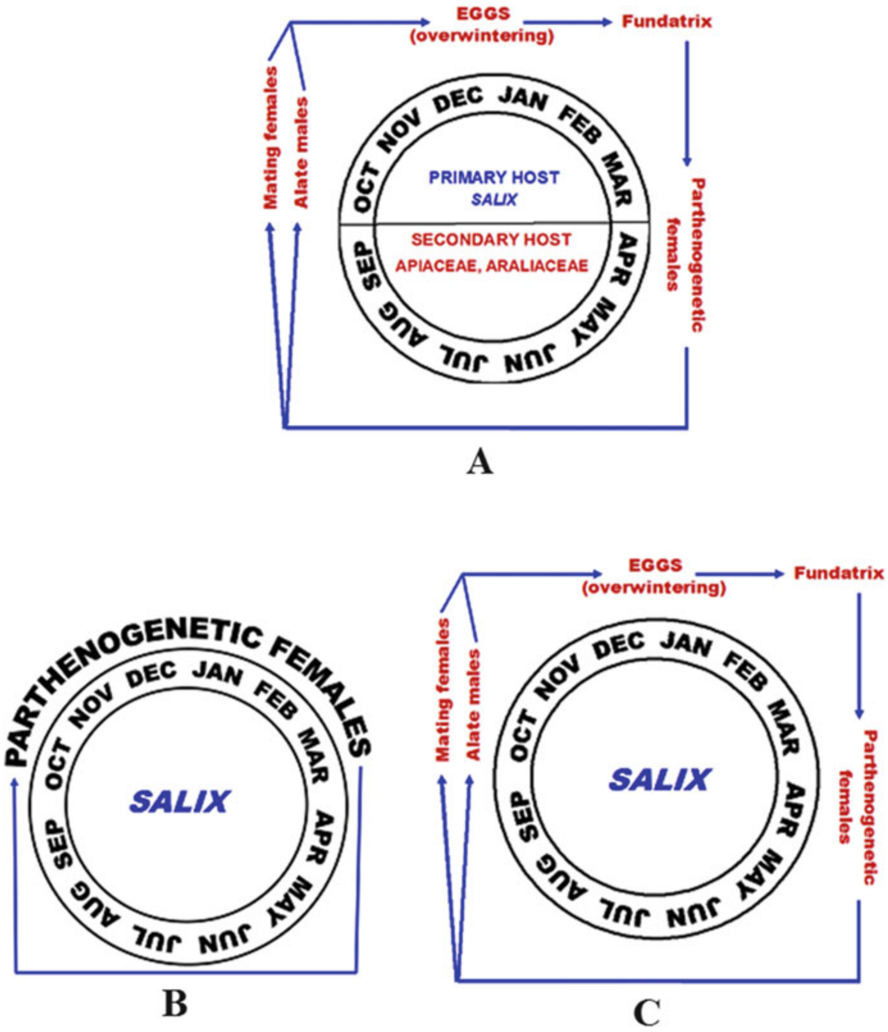


Fig. 3.9 Life cycle pattern of *Salix*-infesting aphids: Host alternating and cyclical parthenogenetic (**a**), non-host alternating and cyclical parthenogenetic (**b**) and non-host alternating and permanently parthenogenetic (**c**) (Medda et al. 1997)

population having the same genotype, exhibited in different generations of the same species. Each morph performs different ecological roles in the life history which is characteristic of aphids (Hille Ris Lambers 1966). This trait coupled with the ability to breed by means of diploid parthenogenesis and viviparity for a major part of the life cycle in aphids has enabled them to produce a large number of clones in different kinds of plants even under adverse conditions (Agarwala 2007). Genetically identical individuals living in different environments may be different in form, physiology or behaviour. Such individuals demonstrate phenotypic plasticity in response to environmental factors like seasonality of their host–plants, food quality, climate and natural enemy association, etc., that vary in space and lime. Agarwala (2007) nicely reviewed this phenomenon in aphids and the factors that cause such variations.

Females may have up to eight genetically identical distinct phenotypes that differ in morphology, physiology, numbers, timing of production, progeny sizes, developmental periods, longevity, host preferences and ability to locate and utilise the alternative host–plants. During the life cycles of a typical migratory aphid, the following sequence of polymorphism is usually met with.

3.2.7.1 Fundatrices: The Stem Mother or Foundress

These are usually apterous, viviparous, viriginoparae or parthenogenetic females which emerge in spring from the overwintered eggs (Fig. 3.10a–c). This morph is characteristic of egg-laying holocyclic aphids. The sense organs, legs and antennae are not so well developed as in succeeding apterous generations; the antennae, for example, are shorter and may comprise a smaller number of segments. The reduction of the parts is apparently correlated with increased reproductive capacity. The eyes are often smaller or consist of fewer facets than in the succeeding generations, and there may be differences in the siphunculi. In *Drepanosiphon platanoides* and some others, the fundatrices are exceptionally alate.

3.2.7.2 Fundatrigeniae or Viriginoparae: Apterous Viviparous Female

These are apterous, parthenogenetic, viviparous females which are the progeny of the fundatrices and live on the primary host (Fig. 3.10d). They are also known as ‘virginoparae’ due to their being virgin mothers, which are prolific breeders under favourable conditions. In heteroecious species where sexual and asexual generations are spent on plants of unrelated taxa, this morph is distinguishable into fundatrigeniae proper and alienicolae. The alienicolae is produced by fundatrigeniae on the secondary hosts.

3.2.7.3 Migrantes: Alate Viviparous Female

The migrantes usually develop in the second, third or later generations of fundatrigeniae and consist of alate parthenogenetic viviparous females (Fig. 3.10e, g). The wings of aptera are sometimes incompletely developed due to local adaptations called brachypterae. The antennae of these morphs are longer than aptera having more sensoria; the eyes are also prominent including ocelli. They develop on the primary host in the beginning of spring, called spring migrants or emigrants, and subsequently fly to the secondary host. In *Drepanosiphon platanoides*, all the viviparous females are winged and consequently fundatrigeniae are wanting. The return migrantes to the primary hosts are the sexuparae or sexuales (Dixon 1998).

Compared to the apterous phenotype, the alate aphids have a longer nymphal development period, lower offspring production and higher longevity (Tsumuki et al. 1990). Moreover, alate aphids are able to tolerate starvation (Hazell et al. 2005). The morphological and physiological characteristics of winged aphids enable

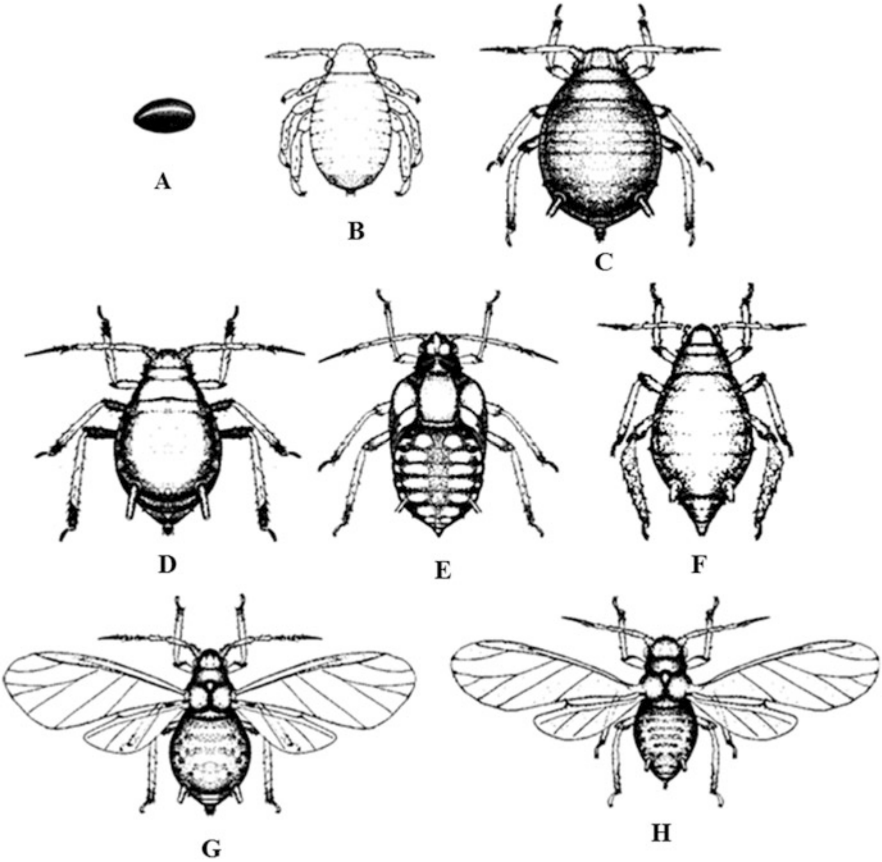


Fig. 3.10 Different morphs of a typical aphid. (**a**) Egg. (**b**) First instar nymph of fundatrix. (**c**) Fundatrix. (**d**) Apterous viviparous female. (**e**) Nymph of alate viviparous female. (**f**) Oviparous female. (**g**) Alate viviparous female. (**h**) Male

them to survive in harsh conditions, have the chance to disperse and clone to a new environment (Dixon et al. 1993).

3.2.7.4 Alienicolae

Alienicolae are parthenogenetic, viviparous females (also known as exule) developing for the most part on the secondary host. They often differ markedly from the fundatrices and migrantes; many generations may be produced comprising both apterous and winged forms.

3.2.7.5 Sexuparae

The sexupara is used to specialised phenotypes that would produce sexual phenotypes in the next generation in holocyclic species (Miyazaki 1987). These are parthenogenetic viviparous females which usually develop on the secondary host, the alate forms migrating to the primary host at the end of the summer. The sexuparae terminate the generations of alenicolae by giving rise to the sexuales.

3.2.7.6 Sexuales

These usually appear only once in the life cycle and consist of sexually reproducing males (androparae) and females (gynoparae), the latter being oviparous (oviparae). The females with rare exceptions are apterous and distinguishable from the apterous viviparous generations of the same sex by the thickened tibiae of the hind legs and the greater body length (Fig. 3.10f). The males are either alate or apterous (Fig. 3.10h). Intermediates between alate and apterous (brachypterous) forms also occur. The sexuales exhibit various types of specialisation among different genera. Apterous parthenogenetic viviparous females may overwinter in several species, e.g. *Brevicoryne brassicae* (Fig. 3.10) and *Myzus persicae*.

With nonmigratory species, the terms migrantes and alienicolae are not applicable. In these cases, the winged and wingless viviparous females are more conveniently referred to as fundatrigeniae alatae or apterae as the case may be, and either one or the other may give rise to the sexuparae.

3.2.7.7 Morphological Changes in Phenotypes

Most of the holocyclic aphids overwinter in egg stage, but in some species, e.g. *Colophina arma*, the apterae viviparae produce very small and stout nymphs, 'midget' that hide themselves in the bark and overwinter without moulting until next spring (Aoki 1980). Similarly, there are several aphid species which aestivate during summer to tide over adverse condition of food and temperature by producing dormant first instar nymph, which remain glued to the leaves until autumn, e.g. *Periphyllus* spp. (Miyazaki 1987). This phenotype has a flattened body covered with plates (Fig. 3.11a) unlike normal ones (Fig. 3.11b).

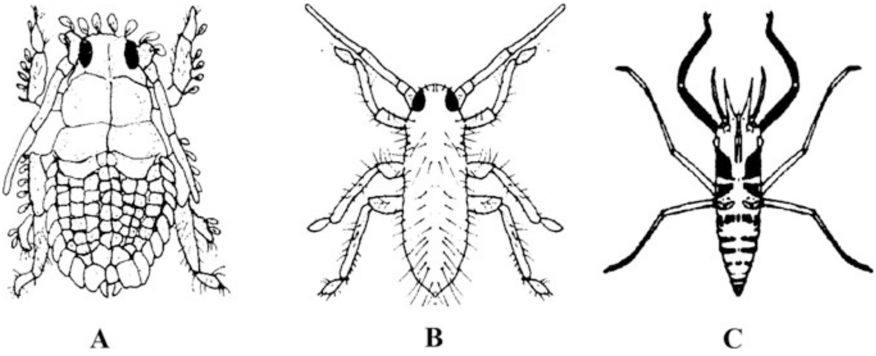


Fig. 3.11 First instar nymph aestivating in summer (**a**) and first instar normal nymph produced in autumn (**b**) of *Periphyllus* sp. infesting acer plants in the Himalayas; sterile first instar soldier phenotype of a bamboo-feeding aphid. *Ceratovacuna* sp. showing frontal horns in the head (Agarwala 2007)

Some species of aphid in divergent taxa, e.g. Eriosomatinae (*Eriosoma, Colopha, Paracolopha, Colophina*) and Hormaphidinae (*Astegopteryx, Pseudoregma, Ceratovacuna*), have evolved 'soldier' phenotype to defend aphid colonies from attacks from enemies and ants (Aoki 1982). However, these morphs do not contribute to reproduction, dispersal, etc., but increase the survival value of parental colonies. Soldiers are not moulting first instar nymphs characterised by sclerotised legs, prehensile forelegs and long and pointed frontal horns.

3.2.7.8 Colour Polymorphism

In colonies of several aphid species, the individuals may be of different colours, such as yellow, brown, red, green, black, pink and purple and various shades of these colours, which match with the coloration of the leaf, flower, fruit and stem of the host–plants on which they feed (Forsman et al. 2008). This affords them certain amount of camouflage. The colour variations may appear irregularly among members of the colony or may be associated with a particular sex or generation (Fig. 3.12). The colour of the aphids is due to a pigment present in their haemolymph, which is derived from the sap obtained from the food plants. The water-soluble pigment known as protoaphin is a glucoside, and the proportion of its constituents determines the various colourations of the aphid. Detail chemistry of the pigments was reviewed by Behura (1996b). Not only do aphids display a bewildering complexity of colour, but their bodies are frequently covered entirely with white or grey wax. In certain species, this wax is secreted only by definite body regions and may be in the form of flakes, ribbons or other shapes. Colour variation is also associated with relative susceptibility to its predators and parasitoids. Müller (1962) had shown that the aphid body colour is genetically determined, with red

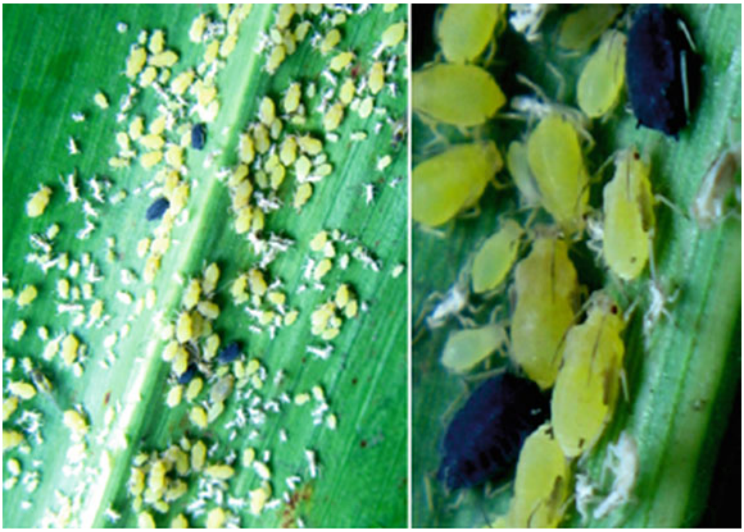


Fig. 3.12 Colour dimorphic (green and black) aphids infesting maize leaf

being dominant over green, and Losey et al. (1997) demonstrated that ladybird beetles tend to devour red aphids on green plants, while the parasitoid wasps preferentially attack green aphids. On these accounts, Losey et al. (1997) concluded that the predation and parasitism pressures appear to regulate the colour variation in natural aphid populations. Moran and Jarvik (2010) discovered that the aphid genome contains several genes for carotenoid synthesis not found in animal genomes and the genes are of fungal origin and seem to have been acquired in the evolutionary history of aphids via ancient lateral transfer. One of the genes is concerned in synthesis of red colour pigments, and its presence or absence is accountable for the red or green coloration of the aphids. Simultaneously, Tsuchida et al. (2010) reported an endosymbiont bacterium, *Rickettsiella viridis*, that modifies body colour of pea aphids in natural populations which is expected to influence prey–predator interactions, as well as interactions with other endosymbionts. Watanabe et al. (2016) demonstrated that ant attendance appears to regulate the proportion of red and green morphs of mugwort aphid, *Macrosiphoniella yomogicola*, in fields. Even the colour polymorphism in aphids appears irregularly among members of the colony or associated with a particular sex or generation.

3.2.7.9 Factors Influencing Morph Determination

There are several factors that influence the aphid to develop into a definite morph. Some of them are associated with their same generation, and others are associated with dispersal. The major factors that determine the gamic forms are the photoperiodism, temperature and the quality of food. In summer season, the factors that always influence aphids to determine their forms are the quality of food, crowding, semiochemicals or pesticides, influence of parasitoids and predators and many other biotic as well as abiotic stresses (Singh and Ghosh 2002). So far as polymorphism is concerned, several factors operate directly or indirectly and interact in various ways (Behura 1994). There are likely to be many different mechanisms, and in only a few species the processes have been analysed experimentally. Agarwala (2007) summarised the factors that could trigger the phenotype plasticity in aphids.

3.2.7.9.1 Photoperiod and Temperature

The photoperiods and temperatures are associated with each other. During shorter photoperiod, temperature is usually low (winter season) and vice versa (summer season). The production of sexual morphs is known in some species to be under photoperiodic control (Dixon 1977; Chen et al. 2019). In *Megoura viciae*, exposure to short-day conditions induces the viviparous females to produce oviparae; there is a critical photoperiod of 15 h at 15 °C, shortening slightly as the temperature rises to a maximum of 23 °C above which the effect does not operate. Photoperiod is perceived directly by the mother (rather than through the host–plant), and this can occur while she is herself developing within the grandmother (Lees 1966). Other species conform to a similar pattern, with differences in the length of the critical photoperiod and of the upper temperature limit, e.g. *Acyrthosiphon pisum* (Lamb and Pointing 1972), *Myzus persicae* (Blackman 1975) and *Rhopalosiphum padi* (de Barro 1992) in which the photoperiod also differs for the production of male and female progeny. Helden et al. (1994) reported a high degree of phenotypic plasticity both within and between morphs of *Sitobion avenae*. Increased production of alate phenotype in response to longer day length and higher temperature has been reported for *Myzus persicae* (Matsuka and Mittler 1978), *Nasonovia ribisnigri* (Diaz and Fereres 2005) and *Rhopalosiphum maidis* (Chen et al. 2019). However, low temperature was reported to induce wing form in *Myzus persicae*, *Lipaphis erysimi*, *Brevicoryne brassicae*, *Aphis glycines* and *Macrosiphoniella sanborni* and high temperature to inhibit wing dimorphism (Lee, 1966; Lv and Chen 1993). Higher temperature also reduced the size of the apterous aphids, e.g. *Myzus persicae* (Tiwari and Singh 2018), as well as their demographic parameters (Pal et al. 2008; Singh and Singh 2015).

3.2.7.9.2 Host–Plant: Food Quality

The cultivated plants vary in species, varieties and cultivars. All such plants have seasonality in development, and content of nutrition varies between crop varieties, growth stages, different parts of the plant, etc. The food quality is one of the important factors that determine the phenotype of aphids (Mittler and Sutherland 1969). Aphids can perceive changes in the quality of their food through gustatory mechanisms and can respond by morphogenetic changes (Mittler 1973). *Aphis gossypii* grown on unsuitable host–plants produces yellow dwarf phenotypes (Watt and Hales 1996). Apterous viviparous phenotype of several aphid species differs in morphometry and growth rates, e.g. *Aphis gossypii* (Singh and Singh 2015a), *Aphis spiraecola* (Dubey and Singh 2008), *Lipaphis erysimi* (Agarwala and Das 1998), *Myzus persicae* (Tiwari and Singh 2016) and *Sitobion miscanthi* (Srivastava and Singh 2008) in response to food plants. *Myzus persicae* produces more apterae on a deficient artificial diet, while *Phorodon humuli* may increase the production of alates when the host–plant grows under unfavourable conditions. *Rhopalosiphum padi* feeding on reduced quality food produce more alate morphs (de Barro 1992). However, Johnson (1966) reported prolonged periods of starvation both of parent aphids and of young nymph induce apterous development in case of *Aphis craccivora*. Secondary metabolites of the host–plants have also been observed to induce alate production (Harrewijn 1978).

3.2.7.9.3 Crowding

Crowding is one of the major biotic factors that induce the production of winged morphs among aphids. Higher aphid densities have been always found to lead to more tactile stimulations between individual aphids, triggering wing induction (Lees 1967; Martínez and Costamagna 2018). Winged morph production has been considered a driver of density regulation in aphids, and in many species, the production of winged individuals is strongly density dependent (Lees 1967; Purandare et al. 2014). However, in few species, e.g. *Metopeurum fuscoviride*, crowding had no effect on the production of winged morphs (Mehrparvar et al. 2013). The production of winged morphs among aphid colonies is crucial in their life history and is the best possible tactic for their dispersal and colonisation in new optimal environments (Müller et al. 2001). However, the stage in the life cycle of the aphid at which crowding has the most influence differs between species (Shaw 1970). The sensitivity of *Rhopalosiphum padi* (Noda 1958), *Therioaphis trifolii* (Toba et al. 1967) and *Brevicoryne brassicae* (Kawada 1965) to crowding seems to be confined almost entirely to the first instar. Thus, crowding tends to promote the appearance of alate virginoparae, though the effect is produced differently in different species. In *Megoura viciae* Buckton, apterous viviparous produces only apterous progeny when reared singly, but when crowded they give rise to alate offspring; the effect is prenatal and not due to nutritional factors (Lees 1966). It is likely that the ultimate causes of the morphogenetic changes that underlie polymorphism are alterations in the endocrine balance during embryonic and postembryonic development.

3.2.7.9.4 Predator–Parasitoid Mediation

The prey phenotypic response to predators is little known. Earlier, it was observed that the magnitude and direction of transgenerational phenotypic responses to predators vary among individuals and/or populations of the same species (Weisser et al. 1999). *Acyrthosiphon pisum* exposed to predator produce offspring developing winged dispersing forms (Dixon and Agarwala 1999; Mondor et al. 2005; Balog et al. 2013; Sentis et al. 2019). The presence of a predator, the larvae of *Chrysoperla carnea*, significantly increased the percentage of winged individuals among the offspring of *Macrosiphoniella tanacetaria*; however, the presence of predators had no effect on the production of winged individuals of *Metopeurum fuscoviride* (Mehrparvar et al. 2013). The antennae of aphids play a major role in perceiving the presence of predators/parasitoids that induce the production of dispersal morph (Kunert and Weisser 2005). Sloggett and Weisser (2002) reported that the parasitoid, *Aphidius ervi*, induced the production of alate morph of *Acyrthosiphon pisum*. Rios Martinez and Costamagna (2017) suggested that these facultative morphological changes may be adaptive as they reduce the probability of predation. Weisser et al. (1999) reported that the kairomones emitted by predators cause plasticity in the morphology, life cycle and behaviour of their prey.

3.2.8 Effects of Inbreeding and Outbreeding

Cyclic parthenogenesis in aphids leads to the peculiar mode of population structures and adaptations to the host–plants. One outcome of this reproductive mode is the frequent incidence of inbreeding; male and female members of the same clone can mate to produce fertilised eggs (Helden and Dixon 1997). This mode of mating is comparable to self-fertilisation in helminths and some annelids. Aphid populations often consist of sexual, asexual and intermediate clones in temperate regions. In such mixed populations, the effects of inbreeding may be restricted. However, a high level of inbreeding may have influenced the local adaptations and the evolution of sex ratios and mating systems in holocyclic populations of aphids. The extent of inbreeding is affected by the life cycle and taxonomic group of the aphid species. As in other insects, inbreeding is expected to occur frequently in species with low migratory ability (Thornhill 1993). In this respect, aphid species without host alternation are likely to inbreed (Komatsu and Akimoto 1995). Furthermore, in the Eriosomatinae and Hormaphidinae, inbreeding may arise easily because the alates of these groups in autumn (sexuparae) have fully grown male and female embryos in their abdomen (Dixon 1998); the sexuals are stout, remain on the host trunk after birth and mate without feeding. Akimoto (2006) observed that when males and females of the same clone of *Prociphilus oriens* are confined in a small cage, they mate readily and produce selfed eggs and, by comparing the hatch rates of selfed and outbred eggs, estimated the effect of inbreeding. These studies indicate that eggs from intra-clonal mating (selfed eggs) hatch less successfully than do eggs from inter-clonal mating, suggesting inbreeding depression. However, the impact of inbreeding depression varied largely among aphid species. Huang and Caillaud (2012) documented the existence of severe inbreeding depression upon selfing in the cyclic parthenogenetic aphid, *Acyrthosiphon pisum*, and opined that the inbreeding avoidance take place sometime between copulation and sperm transfer that suggest that cryptic female choice may play a role in the process. Akimoto (2006) observed that enforced selfing led to a large variation in the hatching time and morphology of first instars. The nymphs hatched out from selfed eggs had longer antennae and tibiae than that hatched out from outbred eggs. Also, their gonads were much smaller in size.

The inbreeding may also influence aphid sex ratios and mating systems. Sexuparae of *Prociphilus oriens* have female-biased sex ratios (Yamaguchi 1985), and Foster (2002) explained it by local mate competition. In the situation where local mate competition occurs, inbreeding is also expected (Hamilton 1967). Akimoto (2006) observed that *Prociphilus oriens* sexuparae consist of two types: one type produces males and females simultaneously in the abdomen (‘M + F’ type), while the other type produces females only (‘F’ type), and the proportions of the ‘M + F’ and ‘F’ types varied from year to year, and accordingly the sex ratio also varied greatly. It is possible that ‘F’-type sexuparae have the advantage of avoiding inbreeding. For understanding the evolution of aphid mating systems, it is necessary to focus on the incidence of inbreeding in the field.

3.2.9 Migration

Johnson (1969) defined insect migration as their periodic flight beyond the boundaries of their old breeding habitats into new ones. Here, the migrants are relatively not distracted during flight by the feeding as well as oviposition stimuli that normally lead. Aphids are important vectors of plant viruses attacking several crops. Therefore, the knowledge of their migration and seasonal presence is necessary to make decisions regarding the time of their control to prevent virus transmission to crops. Aphids fly rather slowly and heavily, but with the help of the wind, they occasionally make astonishing extensive migrations and are capable of very long distance movement. Air current may carry them to altitude of about 1000 m a.s.l. On calm, warm and humid days, thousands of them float in and out among one another, all moving in the same direction by the gentle wind. Most of the aphids are airborne twice a year in India, once in winter (November to January) and the other during spring (March to April). Their aerial activity is minimum during the monsoons (July–August). During winter, the peak hour for alate flight is about noon, while two peak periods are observed in spring, one at noon and the other in less number during afternoon (Ghosh and Raychaudhuri 1980). Several workers have reviewed the ecology of aphid flight (Kring 1972; Parry 2013; Fereres et al. 2017). Most of the aphid migration is related with the search of mates and food and is influenced by a wide range of factors (Johnson 1954).

Pemphigus bursarius is one of the migratory aphid species which occurs on poplar and migrates to the roots of various Poaceae and returns back to poplar in autumn; *Myzus persicae,* in winter, feeds on its primary host, peach, and after winter it migrates to a number of secondary host species (van Emden et al. 1969). *Aphis fabae* overwinters as the egg in autumn on the spindle tree (*Euonymus*), and in May and June, it flies to beans, sugar beet, etc., returning to the spindle tree in October. In Nepal, *Brachycaudus helichrysi* alternates from peach as primary host to *Calendula* and others as secondary hosts. Similarly, *Rhopalosiphum nymphaeae* migrates from the primary host, plum, to the secondary host, water lily. Beginning of the autumn, the sexuparae migrate to the tender leaves of primary host where gynoparae develop which produce oviparous females as well as males. The aphids have potentiality to switch over the plants during unfavourable season on other food plants (Fereres et al. 2017).

3.2.10 Alarm Pheromones: The Defence Chemicals

The sesquiterpene, (E)-β-farnesene, is the alarm pheromone of most of the aphid species (Mondor and Roitberg 2000; El-Sayed 2019). An aphid grabbed by forceps, mimicking a predator, or by an actual predator emits droplets from its siphunculi that contain (*E*)-β-farnesene (Kislow and Edwards 1972; Pickett and Griffiths 1980). Byers (2005) reported that all life stages and sizes of *Aphis gossypii* reared on cotton plants secrete (*E*)-β-farnesene in amounts ranging from 0.1 to 1.5 ng/individual. When any predator arrives the aphid colony, (*E*)-β-farnesene stops aphids from feeding and induces them to escape or drop off the leaf (Wohlers 1982; Pickett et al. 1992). These alarm pheromones also induce the production of wing morph to escape the feeding site for survival (Kunert and Weisser 2005; Kunert et al. 2005; Hatano et al. 2010). Since aphid colonies are composed of genetically identical individuals, therefore, genes for synthesis and recognition of an alarm pheromone in the colony members would increase its complete fitness by warning them to avoid the predator. Few studies have quantified the amounts of (*E*)-β-farnesene in aphids (Vandermoten et al. 2011) and ecological impact of these chemicals on natural enemies of the aphids (Pickett et al. 2017).

3.2.11 Ant Associations

The honeydew of the aphids is the main source of food for many nectar-feeding insects, viz. ants, bees, wasps, parasitoids, dipterans and others. Most of the ants not only lick up the honeydew but also move over the aphid colonies. The aphids produce honeydew more rapidly when the ants tap their antennae on them. In turn, the ants protect the aphids from their natural enemies. Sometimes, the ants carry the aphids from one plant to others if the earlier one begins to wilt. The workers of jet black ant, *Lasius fuliginosus*, carry newly hatched fundatrices of the aphids from the base of oak trees where they overwinter as eggs to new growing leaves at the top of the tree (Ślipiński et al. 2014). The common meadow ants, *Lasius flavus*, even collect aphid eggs in the autumn and early winter and keep them in its nests. In spring, these eggs are transferred nearby the plant roots, so that upon hatching, newly hatched fundatrices find food (Ivens et al. 2012). Some ants are almost aphid dependent for food, while few aphids are obligate myrmecophiles and excrete honeydew only when ants stimulate them to do so. Other ants build roofs of rotten wood over aphid herds that live in cracks in bark; extensions of the tunnels formed in this way are connected directly with the ants' nest. Many ants feed more or less exclusively on honeydew. An amusing sight is an ant helping an aphid to pull its long proboscis out of a plant; evidently, the aphid finds it troublesome to withdraw the rostrum only when some danger requires it to run away as quickly as possible. Sometimes aphids are fed by ants. Flatt and Weisser (2000) observed that the aphids suffer from strong fitness losses if they are not tended by ants.

The number of ants associated with a given species of aphid and the number of aphid species associated with a given species of ant varies from place to place; up to 14 aphid species have been recorded in *Lasius flavus* nests (Depa and Węgierek 2011). Sometimes different ant species which live in similar habitats foster different aphid species, i.e. *Lasius niger* with *Anoecia corni* and *Lasius flavus* with *Anoecia nemoralis*. However, aphid species that have evolved close relationships with ants may have broader environmental tolerances than their hosts; hence, the aphid *Forda formicaria* is attended by *Lasius* spp. in the lowlands and by *Formica* spp. in the highlands (Seckbach and Dubinsky 2011). Ants are not always good with aphids; if aphid numbers increase, the ants used to kill a few off and devour them (Dixon and Hemptinne 2001). Although studies on the mutualism between ants and Indian aphids are scanty, Veeravel and Baskaran (1994) reported that the population of *Aphis gossypii* on brinjal, *Solanum melongena*, is more when the black ant, *Lasius niger*, attended them. Some aphid species are not attended by ants, apparently because their honeydew has a disagreeable quality.

The protection that ants give the aphids they attend is not always excellent and varies depending on the type of predator or parasitoid (Douglas and van Emden 2017). Generally speaking, the ants are better at dealing with ladybird larvae and anthocorid bugs than they are at dealing with lacewing larvae and hover fly larvae. They not only remove the larvae, sometimes killing them, but also remove the eggs of hover flies and ladybirds. Interestingly, by herding aphids onto the tops of the plants, ants render them more vulnerable to attack from some parasitoids (Seckbach and Dubinsky 2011). Detail account of aphid–ant associations has been dealt by Dixon (1985) and Stadler and Dixon (2005).

3.2.12 Endosymbionts

Aphids are frequently engaged in mutualistic associations with bacterial endosymbionts like other insects. Both kinds of symbionts, primary (obligate) and secondary (facultative), occur in aphids.

3.2.12.1 Obligate Endosymbionts

In the obligate relationship, neither aphid nor bacteria is able to survive without the other. Such obligate symbiosis is found in sap-feeding insects like aphids. Aphids feed on the phloem sap of the plants which is an unbalanced diet for them, as it is deprived of essential amino acids. Like other animals, the aphids cannot synthesise these amino acids. For that, aphids harbour certain bacteria symbionts (e.g. *Buchnera aphidicola*) in 60–80 special cells, called bacteriocytes. *Buchnera* is entirely symbiotic and remains viable only inside it (Douglas 1998). This symbiotic relationship was established 200–250 MYA and led to co-speciation of the hosts and their symbionts (Moran et al. 1993). These bacteria are vertically transmitted to eggs and embryos through host generations (Braendle et al. 2003).

3.2.12.2 Facultative Endosymbionts

Facultative symbionts are of two kinds: reproductive manipulators that affect the reproduction of host aphid to maximise their own transmission and the mutualists that can affect a wide range of life history and ecological traits (Oliver et al. 2010; Douglas and van Emden 2017). These symbionts are typically transmitted from mother to offspring, although horizontal transfer occurs at lower frequencies (Gehrer and Vorburger 2012). Among the aphids, *Acyrthosiphon pisum* are the best studied insect–symbiont systems which alone is known to host at least eight different facultative symbiont species (Sandström et al. 2001; Tsuchida et al. 2010). Guo et al. (2017) have described the functions of nine facultative symbionts (*Serratia symbiotica*, *Hamiltonella defensa*, *Regiella insecticola*, *Rickettsia*, *Rickettsiella*, PAXS [pea aphid X-type symbiont], *Spiroplasma*, *Wolbachia* and *Arsenophonus*) of aphids and discussed the associations between these symbionts and aphids, plants and environment. The facultative symbionts grant a wide range of benefits to their hosts, e.g. protection from natural enemies (Oliver et al. 2003; Vorburger et al. 2010), protection from extreme temperatures (Montllor et al. 2002), development of resistance to a fungal pathogen (Scarborough et al. 2005; Łukasik et al. 2013) and the ability to use a greater diversity of resources (Tsuchida et al. 2004). Tsuchida et al. (2011) reported that *Rickettsiella-Hamiltonella* coinfections of *Acyrthosiphon pisum* changed red *aphids* to green. The red/green aphid body colour has ecological, evolutionary and biochemical relevance. In Europe and the USA, red and green aphids commonly coexist within the same populations of *Acyrthosiphon pisum*. It has already been demonstrated that ladybird beetles consume red aphids on green plants (Losey et al. 1997) and that parasitoid wasps *Aphidius ervi* preferentially oviposited into green aphids (Bilodeau et al. 2013), suggesting that these natural enemies with different colour preferences may contribute to the colour polymorphisms in natural aphid populations.

3.3 Aphid–Plant Interaction

Host–plant specificity in aphids varied from extreme monophagy, e.g. *Astegopteryx* spp. on bamboos (Singh and Singh 2018), to highly polyphagy, e.g. the green peach aphid, *Myzus persicae*, whose summer generations develop on about 300 plant species in India (Singh et al. 2015a).

3.3.1 Response to Plant Attributes

The first phase of host–plant selection by aphids is to assess the suitability of the plant as food. For this, the alate aphids use visual and olfactory cues while landing to decide its suitability. Webster et al. (2008) identified chemical cues involved in the long-distance recognition of a host–plant by *Aphis fabae*. Upon landing on the plant, its physical features such as trichomes, simple or glandular, and their density influence the selection behaviour and success of attack of the aphids. Several cultivated crops are resistant against aphids because of these features of the plants. Crops, such as resistant variety of tomato having high density of glandular trichomes, always interfere with the movement and feeding the aphid, *Macrosiphum euphorbiae*. The gustatory receptors located at the back of the mouth help in the recognition of host and non-host–plant. The plants may have inherent toxic chemicals, or upon insertion of the stylet into plant tissue, the non-host–plants secrete toxic chemicals preventing aphid feeding (Schoonhoven et al. 2007). However, few aphids have evolved mechanisms that defend the toxic chemicals present in the plants. For example, brassica crops are rich in glucosinolates (e.g. singrin) to defend insects, but the cabbage aphid (*Brevicoryne brassicae*), mustard aphid (*Lipaphis erysimi*) and *Myzus persicae* evolve mechanisms to defend these chemicals and also use them for host–plant selection (Gols et al. 2008). Similarly, *Macrosiphum euphorbiae* and *Myzus persicae* evolve physiological defence mechanisms from tomatine and solanine (glycosidic alkalosis) of tomato. The glandular trichomes of few plants like *Solanum berthaultii* also secrete (*E*)-farnesene, the aphid alarm pheromone that prevents the colonisation by inducing dispersal behaviour in winged aphis (Gibson and Pickett 1983).

3.3.2 Plant Response to Aphid Attack

Plant responses to aphid attack are different to aphids which are associated or not associated with transmission of viruses. Plant responses to aphids not associated with transmission of viruses are extremely variable. Response of tomato plants to attack of its aphid, *Macrosiphum euphorbiae*, is not evident except the deprivation of plant nutrients making them weak and also susceptible for other insects and pathogens (Guerrieri and Digilio 2008). Similar response of plants to aphid attack can be observed following infestation by generalist aphids, e.g. *Aphis fabae* and *Macrosiphum rosae.* However, in case of *Dysaphis plantaginea*, the injection of aphid saliva may be extremely toxic, leading to localised chlorosis near the feeding site and around the stylet tracks, caused by chloroplast disruption on apple fruits (Miles 1999). Attack of *Aphis spiraecola* on citrus causes growth distortions of its leaves, while peach trees can be curled by *Myzus varians.* The injection of aphid saliva also alters the hormonal balance of the plant, leading to the gall formation that also helps aphid protecting them from their natural enemies and insecticides (Wool 2004).

The aphids transmit phytopathogenic viruses; different kinds of plant responses were noticed after aphid attack. Sometimes, after aphid attack, most virus-infected plants become yellowish in colour that attracts most of the alate aphids. Also, the amount of free amino acids in plant sap is higher in case of virus-infected plants that enhance the development and reproduction of aphids that induces crowding. The crowding induces aphids to differentiate alate morphs that migrate to colonise new healthy plants, thus dispersing the virus. In addition to the above benefit to the aphids that virus infection provides, the plant viruses have an indirect beneficial effect on aphid fitness, by reducing plant defence response. The aphids, in turn, benefit viruses by transmitting them in favourable host for replication; however, few viruses also circulate inside the aphid without replication. Both circulative and replicative viruses make the aphid infective for the rest of its life, with disastrous consequences for agricultural crops (Guerrieri and Digilio 2008).

3.3.3 Indirect Defence Response to Aphid Attack: Tritrophic Context

Singh (2003) reviewed the tritrophic interactions between host–plant, aphids and their natural enemies. These interactions between three trophic levels are physically, chemically and semiochemically mediated responses.

3.3.3.1 Physically Mediated Interactions

It has been demonstrated that the plant architectures influence interactions over several trophic levels. Singh et al. (2000b) have reported that the searching efficiency of *Binodoxys indicus* is highly influenced by foliar pubescence of the food plants supporting its host, *Aphis craccivora* and *Aphis gossypii*. However, sometimes, the physical and chemical influences are not clear, e.g. the decrease in adult survival of the aphid parasitoid, *Aphidius matricarae*, with increasing glandular trichome densities was observed (Obrycki and Tauber 1984).

3.3.3.2 Chemically Mediated Interactions

The direct and indirect effects of plants on herbivores and their natural enemies at the chemical levels have received considerable attention in the literature in recent past. It has been established that after aphid attack, plants release certain volatile chemicals that attract predators and parasitoids of aphids (López Pérez et al. 2007). For example, broad bean plants infested by the pea aphid *Acyrthosiphon pisum* are six times more attractive than uninfested plants towards the parasitoid *Aphidius ervi* Haliday (Guerrieri et al. 1993). In many plant–aphid systems, methyl salicylate is released by aphid infestation that attracts both aphid parasitoids (Sasso et al. 2007) and predators (Zhu and Park 2007). These chemicals are specific for aphid–parasitoid/predator interactions. Singh et al. (2000b) have observed that parasitoid *Binodoxys indicus* parasitises *Aphis gossypii* differently on different host–plants, and Omkar and Pervez (2002) observed the influence of food plants on the predatory potential of an aphid predator, *Coccinella septempunctata.* Infestation of *Prunus persica* by the aphid *Brachycaudus helichrysi* caused a change in the level of different foliar chemicals (soluble sugars and nitrogen, polyphenol, lipid) that provide ovipositional stimulus to the adults of aphid predator, *Eupeodes corollae* (=*Metasyrphus corolla*) (Chakrabarti and Chakrabarti 2002).

3.3.3.3 Semiochemically Mediated Interactions

The plants have an incredibly diverse array of secondary compounds that perform allelochemical functions either as allomones or as kairomones. These chemicals sometimes pass as such into herbivore insect and remain unchanged. Such chemicals or the chemicals synthesised in herbivore insects by modifying the precursors derived from the plants help in their detection by their natural enemies. The literature on host selection by the aphid parasitoids is full of evidence that plant odours attract herbivores as well as their natural enemies (Albittar et al. 2016). The aphid *Brevicoryne brassicae* uses sinigrin (present in brassica plants) as a signal to find host–plants, while its parasitoid *Diaeretiella rapae* uses a related compound allyl isothiocyanate (mustard oil) to find the plant and then the aphid. Therefore, the intrinsic defence of the plant has direct and indirect effects on natural enemies that may be important in biological control and extrinsic plant defence.

3.4 Nature of Damage

Globally, more than 250 species of aphids are pests of both agricultural and horticultural crops (Verma 2000). This figure is only about 5% of the estimated world fauna of over 5110 species (Favret and Eades 2020). Aphids have been reported as one of the devastating insect pests in realising the productivity of many cultivated crops throughout the world. The list of some major aphid pests are displayed in Table 3.2. The damage in some crops is to the extent that nothing remains to harvest such as some cereals, potatoes and rapeseed mustard.

Table 3.2 List of aphid pests of agricultural importance in India and abroad

Aphid species	Plants of economic importance	Name of the virus transmitted
Acyrthosiphon kondoi	Alfalfa	Alfalfa mosaic virus, Australian latent, cucumber mosaic cucumovirus, lucerne and lucerne transient streak virus
Acyrthosiphon pisum	Alfalfa, bean, broad bean, chickpea, clover, cowpea, faba bean, gram, lentil, lucerne, lupin, medics, peas, soybean, snap bean, vetch	Bean common mosaic virus, bean leafroll virus, bean yellow mosaic virus, bean yellow vein banding virus, beet western yellow virus, bidens mottle virus, broad bean severe chlorosis virus, cardamom mosaic virus, chickpea chlorotic stunt virus, chickpea filiform virus, cucumber mosaic virus, faba bean necrotic stunt virus, faba bean necrotic yellows virus, pea enation mosaic virus, pea leafroll virus, pea seed-borne mosaic virus
Amphorophora rubi	Raspberry	Raspberry leaf mottle virus
Aphis (Toxoptera) aurantii	Citrus, coffee, mango, tea, tomato	Citrus tristeza virus, citrus infectious mottling virus
Aphis (Toxoptera) citricidus	Citrus	Cardamom mosaic virus, citrus tristeza virus
Aphis craccivora	Bean, chickpea, citrus, cowpea, faba bean, gram, groundnut, lentil, lupin, medics, pea, pepper, pigeon pea, soybean, snap bean, tomato, vetch	Banana bract mosaic virus, bean common mosaic virus, bean common mosaic virus, bidens mottle virus, cardamom mosaic virus, chickpea chlorotic stunt virus, chickpea stunt disease associated virus, citrus tristeza virus, clover yellows virus, cowpea mosaic virus, cucumber mosaic virus, faba bean necrotic stunt virus, faba bean necrotic yellows virus, garlic mosaic virus, onion yellow dwarf virus, papaya ringspot virus, peanut stripe strain, sunflower mosaic virus, urdbean leaf crinkle virus
Aphis fabae	Artichoke, bamboo, bean, beet, citrus, cucumber, lettuce, pepper, potato, rape, rose, spinach, tobacco, tomato	Artichoke latent virus, bean common mosaic virus, bean yellow mosaic virus, beet mosaic virus, beet western yellow virus, beet yellow virus, cowpea mosaic virus, cucumber mosaic virus, faba bean necrotic yellows virus

(continued)

Table 3.2 (continued)

Aphid species	Plants of economic importance	Name of the virus transmitted
Aphis glycines	Soybean, snap bean	Bean common mosaic virus, cucumber mosaic virus, peanut stripe strain
Aphis gossypii	Brassicas, brinjal, broad bean, celery, chickpea, clover, cotton, cucurbits, faba bean, groundnut, lucerne, pea, pepper, pigeon pea, snap bean, tobacco, tomato	Alfalfa mosaic virus, banana bract mosaic virus, bean common mosaic virus, bean common mosaic virus, bean yellow mosaic virus, beet mosaic virus, cardamom mosaic virus, chickpea distortion mosaic virus, citrus tristeza virus, cowpea mosaic virus, cucumber mosaic virus, dasheen mosaic virus, onion yellow dwarf virus, papaya ringspot virus, pea seed-borne mosaic virus, peanut stripe strain virus, pepper vein banding virus, pepper veinal mottle virus, potato virus, Y, sugarcane mosaic virus, sunflower mosaic virus, urdbean leaf crinkle virus, zucchini yellow mosaic virus,
Aphis helianthi	Sunflower	Potato virus Y
Aphis nasturtii	Cucurbits, groundnut, potato, sweet potato	Potato virus Y
Aphis nerii	Citrus, oleander, papaya, sugarcane	Cardamom mosaic virus
Aphis pomi	Apple, pear, many other fruits	Potato virus Y, plum pox virus
Aphis rumicis	Cardamom	Cardamom mosaic virus
Aphis spiraecola	Citrus, lupin, pepper, potato, tomato, spiraea	Bidens mottle virus, cucumber mosaic virus, potato virus Y
Brachycaudus helichrysi	Artichoke, pigeon pea, potato, tobacco, tomato, some brassicas	Artichoke latent virus, cardamom mosaic virus, large cardamom chirke virus
Brevicoryne brassicae	Cabbage, cauliflower, radish, rapeseed mustard, turnip	Cauliflower mosaic virus, turnip mosaic virus
Ceratovacuna lanigera	Sugarcane	Sugarcane yellow leaf virus
Caveriella aegopodii	Carrot	Carrot mottle virus, carrot red leaf virus
Chromaphis juglandicola	Walnut	Not recorded
Diuraphis noxia	Barley, wheat, other cereal crops	Not recorded
Eriosoma lanigerum	Apple, pear	Not recorded
Greenidia artocarpi	Cardamom, jackfruit	Cardamom mosaic virus
Hyalopterus amygdale	Peaches, plum	Plum pox virus
Hyalopterus pruni	Peaches, plum	Plum pox virus

(continued)

Table 3.2 (continued)

Aphid species	Plants of economic importance	Name of the virus transmitted
Hysteroneura setariae	Maize, paddy, sugarcane, wheat	Bean common mosaic virus, peanut stripe strain
Lipaphis erysimi	Lupin, mustard, rapeseed, turnip	Bean common mosaic virus, bidens mottle virus, turnip mosaic virus
Lipaphis pseudobrassicae	Cabbage, cauliflower, mustard, radish, rapeseed,	Cauliflower mosaic virus, turnip mosaic virus
Macrosiphum euphorbiae	Bean, broad bean, faba bean, lettuce, peas, potato, snap bean, tobacco, tomato	Bean yellow mosaic virus, cucumber mosaic virus, lettuce mosaic virus, pea seed-borne mosaic virus, potato leafroll virus
Melanaphis sacchari	Maize, millets, sorghum, sugarcane	Sugarcane mosaic virus
Metopolophium dirhodum	Barley, millets, wheat	Barley yellow dwarf virus
Myzus ascalonicus	Cabbage, cucumber, garlic, lettuce, onion, turnip	Beet western yellow virus, cucumber mosaic virus, onion yellow dwarf virus
Myzus nicotianae	Tobacco	Papaya ringspot virus
Myzus ornatus	Cucumber, tobacco	Cucumber mosaic virus, potato virus Y
Myzus persicae	Alfalfa, apple, apricot, artichoke, beans, beet, cabbage, celery, cherry, chickpea, clover, cucumber, lettuce, lucerne, lupin, melon, peaches, pears, peas, plum, potato, spinach, tomato, tobacco, turnip	Alfalfa mosaic virus, artichoke latent virus, bean common mosaic virus, bean common mosaic virus, bean yellow mosaic virus, bean yellow vein banding virus, beet mosaic virus, beet western yellow virus, beet yellow virus, bidens mottle virus, cauliflower mosaic virus, chickpea filiform virus, chickpea stunt disease associated virus, citrus tristeza virus, cucumber mosaic virus, cowpea mosaic virus, cucumber mosaic virus, dasheen mosaic virus, garlic mosaic virus, large cardamom chirke virus, lettuce mosaic virus, onion yellow dwarf virus, papaya ringspot virus, pea enation mosaic virus, pea seed-borne mosaic virus, peanut stripe strain virus, pepper vein banding virus, pepper veinal mottle virus, plum pox virus, potato leafroll virus, potato virus Y, sugarcane mosaic virus, sunflower mosaic virus, turnip mosaic virus, watermelon mosaic virus

(continued)

Table 3.2 (continued)

Aphid species	Plants of economic importance	Name of the virus transmitted
Pentalonia caladii	Cardamom	Cardamom mosaic virus
Pentalonia nigronervosa	Cardamom	Banana bract mosaic virus, banana bunchy top virus, cardamom bushy dwarf virus, cardamom mosaic virus, large cardamom chirke virus
Pentalonia kalimpongensis	Cardamom	Cardamom bushy dwarf virus, cardamom mosaic virus,
Rhopalosiphum maidis	Barley, maize, millets, sugarcane, wheat	Banana bract mosaic virus, barley mosaic virus, barley yellow dwarf virus, large cardamom chirke virus, maize dwarf mosaic virus, sugarcane mosaic virus
Rhopalosiphum padi	Barley, maize, wheat	Barley yellow dwarf virus, cereal yellow dwarf virus, large cardamom chirke virus, wheat yellow leaf virus
Rhopalosiphum rufiabdominalis	Barley, maize, millets, wheat	Wheat yellow leaf virus
Schizaphis graminum	Barley, maize, millets, wheat	Cardamom mosaic virus
Sipha flava	Sugarcane, wheat, sorghum	Sugarcane mosaic virus
Sitobion avenae	Barley, millets, sugarcane, wheat	Barley yellow dwarf virus, large cardamom chirke virus
Sitobion fragariae	Maize	Maize dwarf mosaic virus
Sitobion miscanthi	Millets, sugarcane, barley, wheat	Barley yellow dwarf luteovirus, millet red leaf persistent luteovirus
Sitobion rosaeformis	Cardamom, rose	Cardamom mosaic virus
Therioaphis trifolii	Alfalfa, snap bean, yellow clover	Cucumber mosaic virus
Uroleucon compositae	Safflower	Citrus tristeza virus
Uroleucon jaceae	Citrus	Citrus tristeza virus
Uroleucon sonchi	Cardamom	Cardamom mosaic virus
Daktulosphaira vitifoliae	Grape vines	Not recorded

3.4.1 Direct Damage

Aphids attack all parts of the plants including roots. Some of them directly damage the plants by sucking their nutrient that causes a lack of vigour in the plant. The aphid saliva is toxic to plants. Such infested plants have a variety of symptoms such as decreased growth rates, mottled leaves, yellowing, stunted growth, curled leaves, browning, wilting, low yields and ultimately death (Behura and Das 1976). The young seedlings die, the inflorescences fail to blossom and fruits fail to develop normally showing various malformations like twisting of pods, impaired developments of seeds, etc. The subaerial infestations by aphids also cause yellowing of foliages and stunted general growth. Some aphids make different kinds of leaf and stem galls (Chakrabarti 2007). These symptoms are observed on perennial forest trees.

3.4.2 Indirect Damage

In spite of these direct effects, aphids have also some indirect effects. Some species of aphids excrete a high amount of honeydew that covers the stomatal openings hampering their normal physiological processes like photosynthesis, transpiration and respiration. The honeydew also allows the growth of black sooty mould (*Capnodium* spp.) onto the leaves which in turn proves detrimental to the normal activity of plant life. Also, the honeydew has been observed to reduce the effectiveness of fungicides by obstructing their absorption (Dika and Van Pelt 1992). The honeydew also contaminates cotton lint, reducing its quality and economic value. The presence of honeydew on fruit crops can significantly reduce their marketability. Honeydew, as food source, may attract to other crop pest insects. However, the honeydew has some beneficiary act to aphids. It attracts bees, wasps and ants that may provide their protection from their natural enemies such as parasitoids and predators.

The honeydew also provides a valuable food source for beneficial insects such as parasitoids and predators involved in their natural control (Singh et al. 1996, 2000a). The excess honeydew may also nourish soil microorganisms, including nitrogen fixers (Owen and Wiegert (1976). In a nitrogen-poor environment, this could provide an advantage to an infested plant over a noninfested plant (Stadler and Muller 1996), but this does not appear to be supported by the observational evidence (Choudhury 1984).

3.4.3 Damage Through Virus Transmission

Most of the damage caused for the crop by the aphids is their ability to transmit viruses (Eastop 1977). The major viruses transmitted by aphid pests of cultivated plants are given in Table 3.2. Aphid-transmitted viruses account for approximately 50% of the 600 known viruses with an invertebrate vector (Hooks and Fereres 2006). The detail account of transmission mechanisms of aphids as virus vectors was described by Harris and Maramorosch (1977) and Stevens and Locomme (2017). The subfamily Aphidinae includes the majority of aphid vectors (Blackman and Eastop 2000). There are a number of unique features that contribute to the success of aphids as vectors of plant viruses. These include their polyphagous nature (*Myzus persicae*, about 500 plant species worldwide), the ability to undergo parthenogenetic reproduction that facilitates their rapid production and the possession of a needlelike stylet capable of piercing plant cell walls that deliver viruses into host cell. Feeding behaviour and host–plant selection by an aphid affect its potential as a vector (Harris and Maramorosch 1977; Pettersson et al. 2017). The extents to which these factors influence virus transmission depend on the specific virus and its mechanism of transmission (Ng and Perry 2004).

Eight genera of the plant viruses, viz. *Potyvirus*, *Macluravirus*, *Babuvirus*, *Bymovirus*, *Luteovirus*, *Cucumovirus*, *Comovirus* and *Closterovirus*, are transmitted by aphids in India. All three modes of transmission, viz. nonpersistent, semi-persistent and persistent, are observed in India. Nonpersistent virus transmission (stylet-borne) is characterised by very short acquisition and inoculation time in which aphid stylet does not usually pierce beyond the epidermal cells. Because of this, the vectors hardly colonise the host–plant. The aphids begin to lose the infectivity immediately after acquisition. *Alfamovirus, Cucumovirus*, *Fabavirus*, *Macluravirus* and *Potyvirus* are important genera transmitted by aphids in this manner. In semi-persistent, transmission is characterised by few minutes to hours acquisition time with few hours of retention period. *Caulimovirus* and *Closterovirus* are genera transmitted by aphids in this manner. In the persistent-circulative type of transmission, the virus has to be ingested by the aphids and reach the salivary gland via haemolymph. Acquisition period generally ranges from hours to days, and the aphid remains viruliferous for weeks or lifelong. Among the aphid-transmitted viruses, *Luteovirus* and *Babuvirus* are transmitted by aphids in this manner. Ghosh et al. (2017) very nicely reviewed the role of several aphid species as vector of plant viruses of economically important crops.

The green peach aphid (*Myzus persicae*) being highly polyphagous is a vector for more than 110 plant viruses. Cotton aphid (*Aphis gossypii*) transmits more than 75 plant viruses and often infects chilli, potato, sweet potato, brinjal, sugarcane, papaya and groundnuts with viruses. Most of the nonpersistent viruses (e.g. tobacco mosaic virus [TMV]) are transmitted only by aphids. Few semi-persistent viruses (e.g. beet yellow stunt virus [BYV]) and persistent viruses (e.g. potato leafroll virus [PLRV]) are also transmitted by them (Ghosh 1980). A list of virus pathogens transmitted by aphids and aphid species which are regarded as vector of one or more plant viruses has been listed by Ghosh et al. (2017).

The aphids are of great agricultural significance, and nowadays they are being considered as serious pests of agriculture and horticulture. Usually in the absence of primary agricultural crops, i.e. when harvesting of one crop is over, they tide over in unfavourable season on other economic crops; in lieu of these latter crop plants, they just thrive upon many wild plants. Thus, the aphids affect the yield and quality of the seed of several crops which also affect the reproductive potential of the crops.

3.5 Management Measures

3.5.1 Integrated Pest Management (IPM)

Concerns about the risks that the chemical poisons pose to the environment, human health and increased costs of pesticides have increased the urgency for more research into the alternative methods of crop and food protection. The current pest control approaches aim to maximise productivity and approaches that emphasise efficiency and the long-term sustainability of agroecosystems. Indeed, pest management was thought to be a new terminology evolving from pest control to plant protection or crop protection and then pest management and/or integrated pest management (IPM). A broader definition of IPM was given by Stern et al. (1959) as follows: 'Integrated Pest Control is a pest management system that, in the context of the associated environment and the population dynamics of the pest species, utilises all suitable techniques and methods in as compatible a manner as possible and maintains the pest population at levels below those causing economic injury'. Thus, IPM is an effective and eco-friendly approach to pest management that trusts on a combination of common-sense practices and uses current, full information on the life cycles of pests and their interaction with the ecological factors. This approach of pest management is not only cost-effective but also ecologically safe. The IPM takes advantage of all appropriate pest management practices including, but not limited to, the judicious use of pesticides. In practicing IPM, growers should follow a four-tiered approach, viz. monitoring and identification of pests, estimation of economic injury level (EIL) and economic threshold level (ETL), prevention and control measure.

3.5.1.1 Monitoring and Identification

The monitoring and identification of pests removes the possibility that pesticides will be used when they are not really required. The identification of aphid infestation on plants is not difficult. The stunted plants and plants with curled or deformed leaves are likely to have aphid infestations. Feeding aphids usually occur in clusters on succulent shoots, under leaves or in other suitable feeding sites. The presence of honeydew or sooty mould is often an excellent clue that aphids are present. Plants should be examined closely on a regular basis to detect aphids before damage is evident. The evidence of natural enemies such as ladybird beetles, lacewings, syrphid fly larvae, the mummies (parasitised aphids) and disease-killed aphids should be observed. Considerable numbers of any of these natural control means demonstrate that the aphid population may be reduced rapidly without the use of any control practice. Otherwise, some plant protection measure should be applied.

3.5.1.2 Economic Injury Level (EIL) and Economic Threshold Levels (ETLs)

Integrated pest management (IPM) is a well-established strategy for managing agronomically important insect pests (Pedigo et al. 1986) and has been identified as the most cost-efficient tool to reduce aphid outbreaks. The economic injury level

(EIL) and economic threshold level (ETL) are key IPM concepts. Stern et al. (1959) defined the ETL as the density at which control measures should be determined to prevent an increasing pest population from reaching the economic injury level and the EIL as the lowest population that will cause economic damage. The value of ETL is about 75% of the value of EIL. While theoretically there are EIL values for decision-makers to consider in evaluating an ETL, they are largely, if not completely, ignored. That is partly due to the fact that most EIL and ETL differ with aphid species, plant species/varieties, growing stage of the plant, etc.

In other words, the ETL is the population level of insect or extent of crop damage at which the value of the crop damaged exceeds the cost of controlling the pest (Zalom 2010). It is expressed in several ways including the number of insects per plant or per square metre, the amount of leaf surface damage, etc. ETLs have been developed for few aphid species. In the cotton aphid IPM under irrigation conditions, 70% of plants attacked are suggested by Almeida (2001) as the recommended level to start chemical control. In most of the estimates depending on the crop, it ranges from 5 aphids to 300 aphids/plant depending on the plant phenology (Table 3.3). Therefore, EIL and ETL are not fixed values for a pest species, but they varied considerably in relation to the plant growth stages, plant varieties, sowing time and irrigation and also in relation to the cropping seasons. It is essential to know the EIL and ETL of each pest species before control measure on the crop to save the excess cost of pesticide and labour and safe to the other fauna and environment from excess toxicity.

3.5.2 Prevention and Control Measures

High reproductive rate and multiple host sequences provide optimal conditions for aphid population development. The varied habitats, seasonal population development and intra- and intercrop and wild host movement present an extremely complex and difficult challenge requiring new approaches for formulating control and suppression methodology for aphids (Singh 2015). There is really no easy way of controlling these vector insects. In the past, adults were easily killed with insecticides, but insecticide resistance in their populations is a common problem. These insects have become resistant to chemical insecticides quite rapidly, and the wisdom of relying only on chemical insecticides is questioned. Therefore, an integrated approach becomes essential to manage their population. This approach combines the cultural and biological practices with the application of selective insecticides. The cultural control helps in the prevention of aphid infestation on the crop.

3.5.2.1 Mechanical Control

Aphid populations on strong plant may be reduced by knocking them off with a strong spray of water. Most dislodged aphids will not be able to return to the plant, and their honeydew will also be washed off as well. Using water sprays early in the

Table 3.3 Economic injury level (EIL) and economic threshold level (ETL) of some aphid pests

Aphid species	Crop	EIL	ETL	References
Acyrthosiphon pisum	*Medicago sativa*	–	85 aphids/stem	Kulkarni (2016)
	Fenugreek	9.48 aphids/ central shoot	5.18 aphids/ central shoot	Naga and Kumawat (2015)
Aphis craccivora	Cowpea	–	20/2.5 cm shoot length	http://agritech.tnau.ac.in
		16.18 aphids/ central shoot	14.63 aphids/ central shoot	Regar et al. (2016)
	Lablab purpureus	24.9 aphids/ central shoot	21.1 aphids/ central shoot	Godwal (2010)
	Vigna radiata	10–16 aphids/ central shoot	–	Sharma et al. (2000)
	Vigna unguiculata	–	14.63 aphids/ central shoot	Kumawat and Khinchi (2016)
Aphis glycines	*Glycine max*	3.36 aphids/ plants	5 aphids/plants	Xibei et al. (1994)
		–	250 aphids/plant	McCarville et al. (2011)
		674 ± 95 aphids/ plant	273 ± 38 aphids/ plant	Ragsdale et al. (2007)
		–	250 aphids/plant	Seiter (2018)
	KS4202 soybean	526 to 2050 aphids/plant	–	Marchi-Werle et al. (2017)
Aphis gossypii	Citrus	–	25% plants	Pawar (2002)
		271 aphids/m^2	217 aphids/m^2	de Mendoza et al. (2001)
	Cotton	–	30% plants	Stam et al. (1994)
		–	20% plants	Pawar (2002)
Aphis spiraecola	*Citrus clementina*	370 aphids/m^2	322 aphids/m^2	de Mendoza et al. (2006)
		–	45 aphids/ear	Carter et al. (1989)
		–	1–6 aphids/per tiller	Mittnacht (1986)
		–	3–5 aphids/ear	Holz et al. (1994)
		–	4–5 aphids/ear	Shaoyou et al. (1986)
		–	1–8 aphids/ tiller	Larsson (1986)
		–	5 aphids/tiller	Hansen (2003)
Lipaphis erysimi	*Brassica juncea* var. rohini	36.6 aphids/ 10 cm apical twig	34.8 aphids/10 cm apical twig	Saunakiya and Tiwari (2014)

(continued)

Table 3.3 (continued)

Aphid species	Crop	EIL	ETL	References
	Brassica juncea var. vardan	34.4 aphids/ 10 cm apical twig	32.1 aphids/10 cm apical twig	Saunakiya and Tiwari (2014)
	Brassica juncea var. varuna	36.9 aphids/ 10 cm apical twig	Aphids/10 cm apical twig	Saunakiya and Tiwari (2014)
	Canola	–	30-40 aphids/ 10 cm inflorescence	Farooq and Tasawar (2008)
		25 aphids/10 cm shoot	–	Sekhon and Bakhetia (1991)
	Mustard	–	87 aphids/plant	Dubey and Yadu (1998)
		–	88 aphids/plant	Basak et al. (2017)
		20.4 aphids/ 10 cm terminal shoot	15.42 aphids/ 10 cm terminal shoot	Singh and Malik (1998)
Macrosiphum euphorbiae	Oilseed flax	8–10 aphids/stem	–	Wise and Lamb (1990)
		–	3 aphids/stem–flower stage 8 aphids/stem–boll stage	Wise et al. (1995)
Metopolophium dirhodum	Winter wheat	–	10–15 aphids/ear	Wetzel (1995)
Myzus persicae	Chinese cabbage	25 aphids/plant	20 aphids/plant	Jeon et al. (2008)
	Cuminum cyminum	7.15 aphids/three umbels	3.74 aphids/three umbels	Samota et al. (2014)
Nasonovia ribisnigri	Lettuce	–	0.12–0.13 aphids/ plant	Morales et al. (2013)
		–	0.10–0.025 aphids/plant	Nebreda (2005)
Rhopalosiphum padi	*Hordeum vulgare*	–	10 aphids/tiller	Hansen (2000)
	Wheat-booting stage	6.7 aphids/plant	4.7 aphids/plant	El-Heneidy et al. (2003)
	Wheat-growth stage	6.6 aphids/plant	4.4 aphids/plant	El-Heneidy et al. (2003)
	Wheat-heading stage	6.5 aphids/plant	5.1 aphids/plant	El-Heneidy et al. (2003)
	Winter wheat	–	10–15 aphids/ear	Wetzel (1995)
Schizaphis graminum	Wheat-booting stage	5.8 aphids/plant	4.2 aphids/plant	El-Heneidy et al. (2003)

(continued)

Table 3.3 (continued)

Aphid species	Crop	EIL	ETL	References
	Wheat-growth stage	5.7 aphids/plant	4.0 aphids/plant	El-Heneidy et al. (2003)
	Wheat-heading stage	5.9 aphids/plant	4.4 aphids/plant	El-Heneidy et al. (2003)
	Winter wheat	–	< 10 aphids/plant	Robert et al. (1985)
Sitobion avenae	Barley	–	10 aphids/tiller	Larsson (1991)
	Wheat	7 aphids/tiller	1–7 aphid/tiller	Larsson (2005)
	Wheat-heading stage		10 aphids/plant	Li-Jiping et al. (1995)
	Winter wheat		3.5 aphids/ear	Wetzel (1995)
Uroleucon compositae	Safflower	66.4 aphids/5 cm apical twig	49.8 aphids/5 cm apical twig	Anand et al. (2017)
		66.9 aphids/5 cm apical twig	48.8 aphids/5 cm apical twig	Akashe et al. (1997)
		48.1 aphids/5 cm apical twig	38.5 aphids/5 cm apical twig	Kamath and Hugar (2001)
		52.5 aphids/5 cm apical twig	42.0 aphids/5 cm apical twig	Hemagirish et al. (2001)

day allows plants to dry off rapidly in the sun and be less susceptible to fungal diseases.

3.5.2.2 Cultural Control

The cultural control involves changes in crop production practices to make the crop less suitable for the pest or to make it more suitable for the natural enemies or to enhance the ability of the crop to resist pest attack (Norris et al. 2003). Cultural control is an environmentally friendly approach and more of prophylactic in nature than of curative and is frequently first line of defence against pest populations. The following are the important agronomical practices that directly or indirectly affect the aphid biology and keep their population at low level (Sachan 1997; Chang et al. 2017).

3.5.2.2.1 Host–Plant Resistance

The very first event in the farming is the selection of seeds. The seeds of resistant crop varieties should be used for crop production. Crop cultivars resistant to major pests and diseases have been developed in cowpea against *Aphis craccivora* (Omoigui et al. 2017), soybean against *Aphis glycine* (Hill et al. 2004), melon against *Aphis gossypii* (Chen et al. 1997), rice against *Rhopalosiphum padi* (Sun et al. 2017), wheat against *Sitobion avenae* (Hu et al. 2011; Liu et al. 2012), maize against *Rhopalosiphum padi* (Hance et al. 1994) and sorghum against *Melanaphis sacchari* (Sharma et al. 2014) and to a limited extent in pulse and oilseed crops. Host–plant resistance is an efficient and environmentally friendly means of controlling insects, including aphids, but resistant-breaking biotypes have occurred in several plant–aphid systems (Dogimont et al. 2010). There are three mechanisms by which plants become resistant, antibiosis (adverse effect on herbivore biology), antixenosis (induce non-preference behaviour in herbivores) and tolerance (plant traits to withstand herbivore injury), and all these plant traits are regulated by resistant genes (*R*). In recent years, there has been an increase in the knowledge on *R* genes, but only a few *R* genes that cause resistance against aphids have been identified. Some of them include virus aphid transmission (*Vat*) that makes resistance to *Aphis gossypii* in melon (Chen et al. 1997; Martin et al. 2003), recombination-activating gene (*Rag1*) in soybean that provides resistance to soybean aphid, *Aphis glycines* (Li et al. 2007) and *Mi-1.2* gene in tomato that makes resistance to *Macrosiphum euphorbiae* (Linda and Walling 2008). The *Vat* gene in melon enhances the sieve element (SE) wound healing and thus confers resistance to *Aphis gossypii* (Kaloshian et al. 1997). The cloning of *Mi-1.2* gene has been a milestone in plant resistance to aphids (Goggin et al. 2001). Insect-resistant transgenic cotton does not interfere in the performance of aphids (Burgio et al. 2007), but transgenic Indian mustard (*Brassica juncea*) was found resistant against the mustard aphid, *Lipaphis erysimi* (Kanrar et al. 2002).

3.5.2.2.2 Planting Time

This practice is more meaningful if plating of crop is done on the basis of information on the population dynamics of aphid(s) as this is purely based on the phenological asynchrony of the crop with aphid. It is now established that early sown crop either escapes aphid attack or has less degree of infestation. *Brassica campestris* var. *toria* escapes the attack of *Lipaphis erysimi* if sown in mid-September (Sachan 1990). Other brassica oilseed crops suffer less if they are sown between the middle of October to the first week of November depending on the ecoclimatic belt (Upadhyay 1995). Sowing of mustard can be advanced by 10–15 days for escaping from attack of aphids without any appreciable loss in yield (Singh et al. 1984). *Eruca sativa* suffers less due to *Myzus persicae* if planted in October (Singh and Singh 1985). Information is also available on the time of planting of various other crops where they suffer least with aphids. Among the cereals, barley sown between mid-October and mid-November showed less incidence of *Rhopalosiphum maidis* (Singh 1982). Safflower when sown early escapes the attack of *Uroleucon carthami*, particularly at early stage of the crop (Jakhmola 1986). Coriander also suffered less due to *Hyadaphis coriandri* when planted before mid-October (Jain and Yadava 1986). Similarly, lentil planted in early November also showed higher population of *Aphis craccivora* Koch as compared to crop sown in late November or early December (Hossain et al. 2008).

3.5.2.2.3 Manual Removal of Infested Twigs

It is essential to nip the early infestation of aphids in the buds or twigs as aphids after appearance settle on the twigs and multiply from where they disperse to adjoining plants and field. In cotton, removal of the top leaves by hand, using a pruning knife where aphids fed, reduced contamination of bolls below these leaves (Deguine et al.

2000). Plucking and destruction of twigs should be carried out at 15 days interval (Singh et al. 1993).

3.5.2.2.4 Crop Geometry

The primary objective of this cultural method is to maximise yield per unit area without reducing crop quality, so that yield advantages override pest incidence reduction. Plant density has direct influence on the plant growth as well as yield of the crop. Each plant competes with other for nutrients, moisture, sunlight, etc. Dense population may be congenial for some insects, whereas it may be unfavourable to others. Modifying the plant spacing affects the incidence and population development of insect pests in general. A'Brook (1968) demonstrated that *Aphis craccivora* and *Aphis gossypii* were trapped more often over widely spaced than over close-spaced groundnuts. Similarly, in chickpea, wider spacing (60 x 20 cm) or low plant population per unit area resulted in higher population of *A. craccivora* (Lal et al. 1989).

3.5.2.2.5 Intercropping

It includes mixed intercropping, row intercropping, strip cropping, relay cropping and passageway intercropping. Intercropping is preferred over monoculture to avoid risk of crop failure, better utilisation of farm resources and labour and to protect the crop from insect pests. Intercrop reduces the attraction of pest to the host and adversely modifies the microclimate of the pest habitat which may result in impeded dispersal, increased emigration and reduced survival of the pest in the intercrop (Mumford and Baliddawa 1983). Potts and Gunadi (1991) reported a decrease in *Aphis gossypii* populations in potatoes that are intercropped with *Allium cepa* or *Allium sativum*. It has been shown that infestation of *Aphis gossypii* is less in pure crops of green gram, black gram and sunflower as compared to the main crop in combination with cotton. Chamuene et al. (2007) reported that sorghum and pigeon pea intercropped with cotton had fewer *Aphis gossypii*-infested plants and contained abundant population of natural enemies like syrphids, green lacewings and spiders. When beans intercropped with older and taller maize plants interfered with aphid colonisation and only small proportions of beans were infested by the aphid *Aphis fabae* (Ogenga-Latigo et al. 1993). Girma et al. (2000) reported that maize associated with hedge row experienced significantly lower infestation of *Rhopalosiphum maidis* than pure maize. Intercropping of groundnut with pearl millet reduced the incidence of *Aphis craccivora* on main crop (Kennedy et al. 1990). Less population of *Aphis craccivora* was observed on peas when barley and lentil were used as intercrops (Prasad et al. 1987).

3.5.2.2.6 Water Management

Water management is one of the most important factors responsible for proper growth and development of plant and higher yield. Under drought and/or rainfed conditions, plant loses turgidity as well as sap pressure which may result in reduction of feeding, reproduction and survival in aphids. These conditions also stimulate dispersal of aphids. Drought condition increases the solute concentration and sap viscosity to such an extent that feeding by aphid is drastically hampered (Bakhetia and Brar 1988). Population of *Lipaphis erysimi* increases on mustard crop, *Brevicoryne brassicae* on cabbage and *Aphis craccivora* on lentil and groundnut under irrigated conditions. Mustard crop should be irrigated twice to avoid heavy aphid infestation (Gangasaran and Giri 1986). Samuel et al. (2006) observed that short watering intervals (regimes) increase the population of *Aphis craccivora* on cowpea. Regarding water management in various crops, under no irrigation, *Brassica carinata* suffered heavily and succumb to injury of *Lipaphis erysimi* (Bakhetia and Brar 1988). On the contrary, Prasad et al. (1987) could not find any difference in the population of *Aphis craccivora* and *Acyrthosiphon pisum* on irrigated and unirrigated peas.

3.5.2.2.7 Fertility Management

There are 20 essential plant elements which are needed for the growth and development of the plants (Barker and Pilbeam 2016). Out of these, N, P and K are major nutrients. In general, high nitrogen supply results in increased tissue softness and water content as carbohydrates making the plant more susceptible to attack by aphids (Nevo and Coll 2001). Excess nitrogen in the plant limits proteolysis which results in poor nitrogen level in sap, whereas poor level of soil nitrogen leads to reduced concentration of soluble amino acids and amides in the plants (Tingey and Singh 1980). High dose of nitrogen increases the population of *Lipaphis erysimi* (Sidhu and Kaur 1977; Singh et al. 1995), *Aphis craccivora* (Sridharan et al. 1990) and *Myzus persicae* (Kashyap and Bhanot 1987). The presence of higher level of phosphorus makes the plant less susceptible for aphids. However, potassium has balancing effects on nitrogen and phosphorus. Deficiency of potassium results in the accumulation of soluble nitrogen and carbohydrates owing to inhibition of protein synthesis and increase in the rate of proteolysis (Tingey and Singh 1980). The presence of potassium causes toughening of plant tissues which might be due to decrease in protein by corresponding increase in carbohydrate content. In general, P and K application decreased the population of *Lipaphis erysimi* on mustard (Singh et al. 1995). Decreasing level of K led to greater fecundity of *Myzus persicae* and *Brevicoryne brassicae* on brussels sprout (van Emden 1966). The lowest aphid population was noticed at 80 kg/ha of DAP. Higher proportion of N:P:K (80:40:30) showed higher population of *Lipaphis erysimi*, whereas 40:80:40 ratio reduced aphid infestation (Singh et al. 1995). Similarly, high N:P:K (225:90:45) increased population of *Brevicoryne brassicae* on cauliflower (Sinha et al. 2018).

3.5.2.2.8 Removal of Alternate Hosts

Important aphids like *Myzus persicae, Aphis craccivora* and *Aphis gossypii* are polyphagous in nature and thrive well on cultivated as well as on wild plants. These wild plants and weeds provide suitable habitat and food for the aphid during off season. Wheatgrass, *Agropyron cristatum*, and Canada wildrye, *Elymus canadensis*, were observed as alternative hosts of the Russian wheat aphid, *Diuraphis noxia*, and serve as hosts between the time winter wheat was harvested and planted (Armstrong et al. 1991). Removal of such plants prevents the initial population ready for attack on the main crop, wheat. Destruction of stray groundnut plants and weeds has been recommended for the management of *Aphis craccivora.* Similarly, the destruction of yellow flowering weeds has been found useful against *Myzus persicae* in potato field (Raman 1985).

3.5.2.2.9 Trap Crop

Trap crop is generally used to ward off the insects from the main crop. It prevents the insects from reaching the main crop. Trap crop is more attractive and susceptible than the main crop. The planting of trap crop is done in such a time that its susceptible stage coincides with peak activity of the insect. Mustard as trap crop has been found very useful in the management of *Lipaphis erysimi* and *Brevicoryne brassicae* on cabbage when planted in mustard/cabbage (2:9) ratio (Srinivasan and Krishana Moorthy 1991). Firstly, it can attract aphids and draw them away from their host–plants. Secondly, it can alter the recognition of the host–plant. This effect is mostly attributed to companion plant volatiles since they disturb the aphid host–plant location, and additionally they may react chemically and physiologically with the host–plant, making it an unsuitable host for aphids. Thirdly, it can attract natural enemies by providing shelter and food resources (Ben-Issa et al. 2017).

3.5.2.2.10 Distance from Other Crops

Closely related or crops grown for different purposes should be planted distantly so that insects from one crop may not be able to reach other crop where physiological conditions suitable for aphid deteriorate. *Toria* and *Sarson* should be sown away from mustard and other long duration brassicas. Seed plot of potato should be away and located upwind from the commercial potato (Raman 1985).

3.5.2.2.11 Rogueing and Avoidance of Ratooning

Rogueing of aphid infested plants and avoidance of ratooning have been found very useful in the management of banana aphid, *Pentalonia nigronervosa*, a vector of bunchy top virus, and *Aphis gossypii* which transmits cucumber mosaic virus (Tandon 1994).

3.5.2.2.12 The Use of Reflective Mulches

Reflective plastic mulch, or silver-coloured plastic mulch, is covering material placed in fields when the plants are young and initially aphid-free. This mulch reflects light that interferes with the ability of flying aphids to locate plants which delay or reduce the extent of infestation of young plants by winged aphids (Stapleton and Summers 2002). It works better for small horticultural and vegetable crops that are especially sensitive to viral diseases transmitted by aphids. As plants grow larger, reflective mulch becomes less effective. Reflective mulch ceases to repel insects by the time the plant canopy covers more than about half of the soil surface. In addition to above benefit, the reflective mulch also enhances growth of the plants by increasing photosynthesis and reducing heat and water stress by keeping the plant and soil cooler (Pramanik et al. 2015).

3.5.2.3 Biological Control

Biological control of aphids in the fields has been successfully achieved in several parts of the world because their predators and parasitoids have great potential in managing their populations in spite of certain limitations. Indeed, biological control has been a central core around which IPM has commonly been developed (Dent 2000). The reason for this is that natural enemies constitute the major natural control factors, which can be manipulated. Singh (2001), Joshi et al. (2010), Boivin et al. (2012), Singh and Singh (2016) and Hance et al. (2017) have given a detailed account of biological control of aphids in India and abroad. In addition, entomopathogenic fungi have also been found to control few aphid species (Meyling and Eilenberg 2007).

3.5.2.3.1 Predators

Aphid predators belong to four orders of insects: Coleoptera (families Coccinellidae and Carabidae), Diptera (families Chamaemyiidae, Syrphidae and Cecidomyiidae), Hemiptera (families Anthocoridae and Geocoridae) and Neuroptera (family Chrysopidae). Ladybird beetles are most common aphid predators encountered throughout the world (Fig. 3.13a and c). The common genera predaceous on aphids are *Adalia*, *Adonia*, *Brumoides*, *Coccinella*, *Cheilomenes*, *Exochomus*, *Hippodamia*, *Oenopia*, *Micraspis*, *Scymnus*, etc. Aphidophagous coccinellids have a long history of importation in classical biocontrol with only few recognised successes (Obrycki and Kring 1998). Dixon (2000) judged only one to be substantially successful after

Fig. 3.13 Predators of aphids. Adult ladybird beetle (**a**), hover fly larva (**b**), ladybird larva (**c**) and common green lacewing larva (**d**)

155 tallied intentional introductions of coccinellid species worldwide that specifically targeted aphids. The aphid midge, *Aphidoletes aphidimyza*, is a cecidomyiid fly whose larvae are effective predators of aphids, an important component of biocontrol for greenhouse crops (Boulanger et al. 2019), and is commercially available (e.g. APHIDEND®, Koppert B.V., The Netherlands). Several species of syrphid flies have been evaluated as biocontrol agents against aphids (Fig. 3.13b) (Joshi and Ballal 2013), and few species are commercially available, e.g. *Episyrphus balteatus* (SYRPHIDEND®, Koppert B.V., The Netherlands). Green lacewings, particularly members of the genera *Chrysopa*, *Chrysoperla* and *Mallada* (Chrysopidae) (Pappas et al. 2011) (Fig. 3.13d), and brown lacewing (Hemerobiidae) are major biocontrol agents of aphids among Neuroptera and have been used against aphids in several parts of the world (Rocca and Messelink 2017).

3.5.2.3.2 Parasitoids

Most aphid parasitoids belong to Hymenoptera (Braconidae, Fig. 3.14, and Aphelinidae). Most of the aphidiine parasitoids used in biocontrol belong to the genera: *Aphidius*, *Binodoxys*, *Diaeretiella*, *Ephedrus*, *Praon* and *Trioxys*. They are cosmopolitan in distribution so that one species, *Diaeretiella rapae*, parasitises about 98 species of the aphids distributed in 87 countries throughout the world

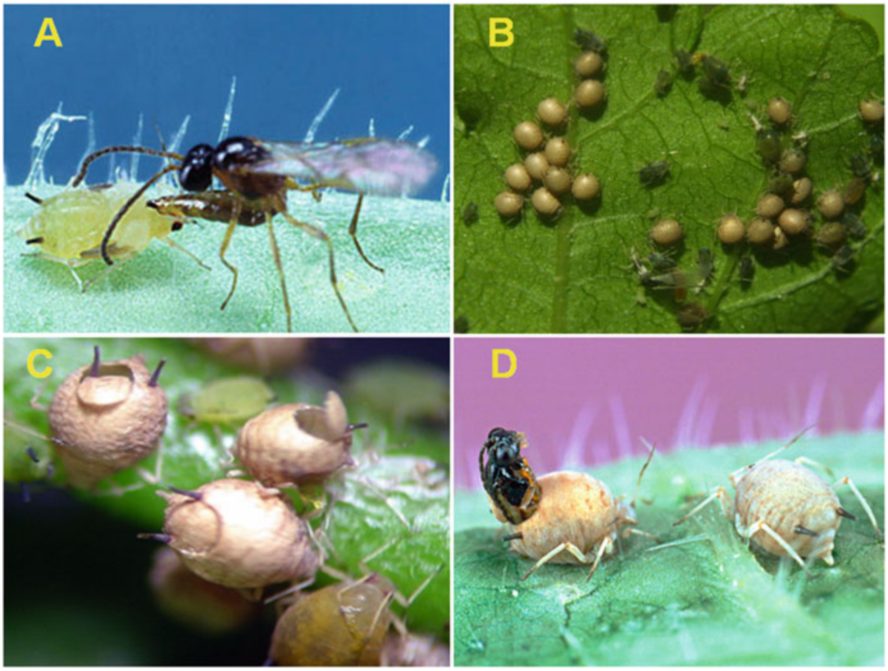


Fig. 3.14 A female parasitoid parasitising an aphid (**a**), parasitised aphids as "mummies" (**b**), mummies with emergence hole with cap (**c**), adult parasitoids after making a hole emerging out the mummy (**d**)

(Singh and Singh 2015b). More than 100 biocontrol programmes have been monitored against at least 30 species of aphids, and about 50% of them proved successful. These programmes include the introduction of about 25 species of parasitoids. Singh and Singh (2016) enlisted and summarised major biocontrol attempts against the aphids throughout the world. The parasitoids become established in 34 out of 57 attempts. The introduction of *Aphelinus mali* in France to control the woolly aphid, *Eriosoma lanigerum*, in apples was probably the first attempt of biocontrol of aphids (Howard 1929). It was then introduced in several European countries, Australia, New Zealand and India. The Indian species *Aphidius smithi* quickly established in Mexico, Canada and the USA in the fields of alfalfa (Mackauer 1971). Similarly, *Aphidius eadyi* successfully controlled the pea aphid, *Acyrthosiphon pisum*, in New Zealand, and *Trioxys complanatus* and *Trioxys tenuicaudus* suppressed the population of alfalfa aphid, *Therioaphis trifolii*, and elm aphid, *Tinocallis platani*, respectively, in the USA (Hughes 1989). Active biocontrol attempts have been made by the introduction of *Diaeretiella rapae* against the Russian wheat aphid *Diuraphis noxia* with partial success (Brewer and Elliott 2004). Singh and Agarwala (1992) and Singh and Rao (1995) demonstrated successful control of *Aphis craccivora* on pigeon pea and *Aphis gossypii* on cucurbits by introducing the indigenous parasitoid *Binodoxys indicus*. Levie et al. (2005) showed that the release of 20,000 *Aphidius rhopalosiphi* per hectare in wheat crops, twice at 1-week intervals, allowed the control of the aphid, *Sitobion avenae*. In China, mass release of *Aphidius gifuensis* was used to control *Myzus persicae* in tobacco crops (Yang et al. 2009). In apple orchards, the inundative release of two parasitoid species, *Ephedrus persicae* and *Aphidius matricariae*, controlled the population of rosy apple aphid, *Dysaphis plantaginea* (Boivin et al. 2012). Waterhouse (1998) summarised the attempts of biocontrol of *Aphis craccivora* and *Aphis gossypii* using their parasitoids in several countries such as Australia, China, Columbia, Cuba, East Asia, France, India, Iraq, Israel, Italy, Japan, Korea, Malaysia, Netherlands, Pakistan, Philippines, USA, Russia, Vietnam, etc.

Glasshouse crop cultivation is a striking example of recent development in the field of biocontrol. Around 55 years ago, even specialists had serious doubt about the success of biocontrol in the glasshouses because this method of crop raising is economically vulnerable. Parr and Scopes (1970) described the problems associated with biocontrol of glasshouse pests. According to them, biocontrol gives more predictable control lasting several weeks to months despite being cheaper and eco-friendly. Paprikas, tomatoes, lettuces, chrysanthemums and other ornamental pot plants are cultivated in glasshouses mostly in Europe. All these plants severely suffer with *Myzus persicae*. Successful biocontrol of *Myzus persicae* was achieved by introducing *Aphidius matricariae* (Hussey and Scopes 1985) and *Ephedrus cerasicola* (Hofsvang and Hågvar 1980; Hågvar and Hofsvang 1990). Biocontrol through inundative or inoculative releases is applied in greenhouses where it gives the best results (van Lenteren 2000). During the year 2006, more than 37,000 ha of greenhouses was under biocontrol programmes (Parrella 2008). However, the augmentative use of parasitoids for aphid biocontrol requires the release of thousands of individuals. For instance, under greenhouse cultivation, quantities of parasitoids

Table 3.4 Attempts and success of biocontrol of *Myzus persicae* in glasshouses using its parasitoids

Aphid species	Parasitoid species	Crop	Country	References
Myzus persicae	*Aphidius colemani*	Cucumber	Netherlands	van Lenteren et al. (1997)
	Aphidius matricariae	Cucumber	Netherlands	van Lenteren and Woets (1988)
		Brinjal	France	Rabasse et al. (1983)
		Chrysanthemum	UK	Wyatt (1985)
		Sweet pepper	UK	Buxton et al. (1990)
		Sweet pepper	Netherlands	Ramakers (1989)
		Vegetables, ornamentals	Germany	Albert (1990)
		Sweet pepper	USSR	Popov et al. (1987)
		–	Bulgaria	Loginova et al. (1987)
		Tomato	Canada	Gilkson (1990)
		–	Hungary	Polgar (1987)
	Aphidoletes aphidimyza	Vegetables	Finland, Denmark, Canada, USA, Russia	Markkula and Tittanen (1985)
	Ephedrus cerasicola	Sweet pepper	Norway	Hofsvang and Hågvar (1980)
Myzus ascalonicus	*Aphelinus abdominalis*	Strawberry	Denmark	Sigsgaard et al. (2013)
	Aphidius colemani	Strawberry	Denmark	Sigsgaard et al. (2013)
	Aphidius ervi	Strawberry	Denmark	Sigsgaard et al. (2013)
	Aphidoletes aphidimyza	Strawberry	Denmark	Sigsgaard et al. (2013)

released for aphid control range from 2500 to 10,000 individuals per ha (van Lenteren 2003). Table 3.4 summarises the attempts and success of biocontrol of *Myzus persicae* and *Myzus ascalonicus* in glasshouses worldwide.

3.5.2.3.3 Pathogenic Fungi

The entomopathogenic fungi are different kinds of biocontrol agents of insect pests (Rehner 2005). Twenty-eight mycopesticides using seven species of entomopathogenic fungi, such as *Beauveria bassiana, Metarhizium anisopliae* and *Lecanicillium* spp., are commercially available in several countries for control of aphid pests (Goettel et al. 2005; de Faria and Wraight 2007). Sabbour (2019) demonstrated the promising control of *Myzus persicae* by using destruxins which is isolated from *Metarhizium anisopliae*. It is a cyclic hexadepsipeptide and causes paralysis and a speedy death to the insects, and also it causes suppression of the insect immune system. The destruxins are also used as synergist to enhance the efficiency of other biopesticides used against aphids (Yi et al. 2012).

These mycopesticides mainly use propagules such as conidia, blastospores or hyphae which have advantages of direct mortality of the target aphid pest (Shan and Feng 2010). However, the conidia of entomopathogenic fungi are highly affected by environmental factors, such as temperature and relative humidity, and are slow in causing mortality. These factors have prevented wider application and use of these biocontrol agents. In spite of that, these species are being applied in agriculture and forestry in temperate regions (Meyling and Eilenberg 2007; Kim et al. 2013). However, these pathogens adversely affect the potential of aphid parasitoids. For example, *Beauveria bassiana* have been found to reduce the emergence and longevity of females of *Diaeretiella rapae* against *Myzus persicae* (Silva et al. 2014). Moreover, González-Mas et al. (2019) could not observe any detrimental effect on the predator *Chrysoperla carnea* and the parasitoid *Aphidius colemani* of *Aphis gossypii* on melon crop when inoculated by *Beauveria bassiana* in Spain. Therefore, application of these two kinds of biological control agents in combination of the control of aphids requires effective time management to avoid antagonistic interactions. The use of entomopathogenic fungi in the management of insect pests of field crops has been reviewed by Maina et al. (2018).

3.5.2.3.4 Bacteria

The biosurfactants produced by *Bacillus atrophaeus* L193 were reported to control the aphid, *Rhopalosiphum padi,* in order to suggest a friendly alternative to chemical pesticides. These surfactants contain lipopeptides, such as surfactins, fengycins, bacillomycins and iturins, that cause aphid death by affecting cuticle membranes (Rodríguez et al. 2018). A multifunctional endophytic bacterial strain *Bacillus velezensis* YC7010 has been found to induce systemic resistance against bacterial and fungal pathogens of rice. Rashid et al. (2017) demonstrated that root drenching of the brassica plant *Arabidopsis* seedlings with this bacterial strain induces higher accumulation of hydrogen peroxide, cell death and callose deposition in leaves that reduced settling, feeding and reproduction of *Myzus persicae* on its leaves. Foliar spray of a bacterium, *Pseudomonas poae*, to plants before aphid colonisation was found to reduce the infestation by *Myzus persicae* (Paliwal 2017). Similarly, field application of another species, *Pseudomonas fluorescens*, was observed to control *Aphis gossypii* on *Bt* and non-*Bt* cotton (Manjula et al. 2018).

3.5.2.3.5 Interactions Between Host–Plant Resistance and Biological Control

The development of resistant crop cultivars offers a sound and very practical approach to the long-range control of certain agricultural pests. However, the varietal breeding is traditionally in the botanical field of plant breeding that falls rather outside the scope of the biological control workers. Recent advancement of our knowledge about its compatibility with biological control necessitates interdisciplinary action in achieving good pest management. Nonlethal plant defences may potentiate the role of predation, parasitism and pathogen infection in regulating herbivore population. It is usually assumed that host–plant resistance is generally or highly compatible with integrated pest management and biological control strategies (Kogan and Jepson 2007). Host–plant resistance can affect the natural enemies of insects in several ways. In some situations, plant resistance can reduce the performance of natural enemies, while in others it can act in a complimentary way (Price 1986). For example, while aphids feed on plants, plants produce semiochemicals, which may act as repellents to aphids or attractants to natural enemies of these aphids (Biswas and Singh 1998). Presumably, this control results because the resistant variety facilitates the searching behaviour of the enemy, reduces the vigour of the host to avoid parasitisation, delays development of the host so that the pest and the enemy populations are temporarily synchronised and/or among other things modifies the behaviour of the host so that it is more easily parasitised (Price et al. 1980).

Each and every plant defends themselves by herbivore insects in general either by producing chemicals, such as toxin, or digestibility reducers, or through physical defence by trichomes or toughness, or by a combination of the two, as with glandular trichomes or resins (intrinsic defence of the plants, resistance of host–plant), and by benefiting natural enemies of the herbivores (extrinsic defence of the plants) (Singh 2003). It is now recognised that almost every mechanism of the intrinsic defence of a plant has an effect on the trophic system and that intrinsic defence may impact positively or negatively upon the third trophic level as well as on those factors involved with extrinsic defence (Price et al. 1980; Singh et al. 2000b). The intrinsic and extrinsic defences of plants reduce the colonisation rate of the herbivores.

Plant trichomes are one of the bases of host–plant resistance against a number of aphids. The presence of trichomes in potatoes confers resistance against the aphid *Myzus persicae* and *Macrosiphum euphorbiae* (Tingey et al. 1982).

Du et al. (1998) observed that the plant *Vicia faba* synthesises some chemicals particularly 6-methyl-5-hepten-2-one after the infestation by the pea aphid *Acyrthosiphon pisum*, and this chemical was observed most attractive for its parasitoid *Aphidius ervi*. Price et al. (1980) suggested the melding of these two approaches is essential in evaluating the roles of natural enemies in population control of herbivores. Several workers have studied the resistance of cereal crops to a variety of pests, particularly to biotypes of *Schizaphis graminum*. The synergistic ability of host–plant resistance with the parasitoid *Lysiphlebus testaceipes* was demonstrated by several workers in the past (Burton and Starks 1977; Starks and Burton 1977). They have observed that greater degree of parasitism by *Lysiphlebus testaceipes* on resistant variety of sorghum and oats resulted from increased degree of movement of the aphids, which made them several times more susceptible to parasitisation. They observed that resistant varieties of sorghum and oats enhance the killing efficiency of the parasitoid *Lysiphlebus testaceipes* in reducing the population of the aphid *Schizaphis graminum*. Biswas and Singh (1998) observed increased efficiency of *Lysiphlebus delhiensis* against *Melanaphis sacchari* on resistant corn cultivar. Fuentes-Contreras and Niemeyer (1998) observed that hydroxamic acids, plant secondary metabolites associated with aphid resistance in wheat, influence the host acceptance and suitability of the aphid, *Sitobion avenae*, to its parasitoid *Aphidius rhopalosiphi*. Similarly, Fuentes-Contreras and Niemeyer (2000) reported significant reductions of population growth rate of aphids with the joint action of wheat resistance and its natural enemies. Farid et al. (1998a, b) observed that the plant resistance against the Russian wheat aphid, *Diuraphis noxia*, did not have an adverse affect on the percentage of its parasitism by *Diaeretiella rape*.

Cultural control practices may also be integrated with biological control and host–plant resistance (van Emden 2017).

3.5.2.3.6 Interaction Between Cultural Control and Biological Control

Increasing the diversity within the agricultural fields by introducing multiple cropping, intercropping, strip harvesting, selective retention of weeds within the crop or conservation of wild plants at field margins also promotes the conservation of natural enemies of aphids and also enhances their natural control ability. Increasing diversity within crops is predicted to provide a greater number of opportunities for natural enemies to survive in agricultural systems and also tends to increase natural enemy abundance and diversity, providing a system more resilient to pest population increase (Rodriguez-Saona et al. 2012). Vegetational diversity also provides support for insect biocontrol at local and landscape levels. The plants serve as a reservoir of the alternative host species, and flowering plants are important sources for food as the adult parasitoids do not necessarily feed only on honeydews but also on pollen, nectar and other sugary plant secretions.

Provision of food resources, such as floral (nectar and pollen), extrafloral (nectaries, exudates and fruits) and insect products (honeydew) in the fields, had significantly increased the longevity and potential fecundity (egg load) in the aphid parasitoids *Aphidius rhopalosiphi* (Budenberg et al. 1992), *Diaeretiella rapae* (Tylianakis et al. 2004), *Aphidius colemani* (Charles and Paine 2016) and *Aphidius ervi* (Hogervorst et al. 2007); searching activity of *Aphidius nigripes* (Bouchard and Cloutier 1984) and *Lysiphlebus testaceipes* (Grasswitz and Paine 1993); and intrinsic rate of increase of *Lysiphlebia mirzai* (Singh et al. 1996) and *Lipolexis scutellaris* (Singh et al. 2000a). Costello and Altieri (1995) reported that *Diaeretiella rapae* highly parasitised *Myzus persicae* on clean cultivated broccoli, and Shlyakhovoi and Bobonich (1975) observed that parasitism of *Brevicoryne brassicae* on cabbage by *Diaeretiella rapae* was high if nectar-bearing plants are grown in its neighbourhood. These positive effects on the parasitoid's reproductive activity improve its effectiveness of both conservation and augmentation biological control of aphids.

3.5.2.3.7 Interaction Between Transgenic Plants and Biological Control

Crop plants transformed to express toxin genes derived from *Bacillus thuringiensis* (*Bt*) provide high levels of resistance to certain pest species, which is likely to have consequent effects on parasitoids specialising on such pests. A better understanding of the interaction between transgenic plants, pests and parasitoids is important to limit disruption of biological control and to provide background knowledge essential for implementing measures for the conservation of parasitoid populations (Singh

2003). It is also essential for investigations into the potential role of parasitoids in delaying the buildup of Bt-resistant pest populations. Introducing genetically modified insect-protected crops into the agricultural landscape has a profound effect on target herbivore abundance and distribution. Populations of specialised natural enemies are expected to be reduced because vast acreages of crops will no longer contain appropriate hosts. However, hosts should still be abundant in refuge plantings designed to prevent the spread of resistance in the target herbivore populations.

Behavioural choice tests with maize expressing the Cry1Ab toxin of *Bt* and larvae of the predatory lacewing *Chrysoperla carnea* demonstrated that the predator preferentially feeds on aphids rather than on lepidopteran larvae (the targets of the *Bt* toxin) (Meier and Hilbeck 2001). This preference will reduce the exposure of *Chrysoperla carnea* to Cry1Ab toxin since aphids do not ingest the toxin when feeding on *Bt* maize (Raps et al. 2001). Population-scale laboratory experiments with the aphid *Myzus persicae* and its parasitoid, *Diaeretiella rapae*, showed that *the parasitoid* was as effective in controlling this nontarget pest on *Bt* and proteinase inhibitor oilseed rape plants as on untransformed plants (Schuler et al. 2001).

3.5.2.4 Chemical Control

As far as possible, chemical control of aphid pests should be avoided as most of them are fatal for honey bees and other pollinators and other beneficial insects, particularly the parasitoids and predators of aphids. Only when the use of insecticide becomes inevitable, then insecticides may be used to control the aphid population in different agroecosystems (Dewar and Denholm 2017). The main aim with insecticide use should be to select such insecticides that have minimal detrimental effect on pollinators and natural enemies but are still effective on the insect mortality.

According to the crop, multiple applications of insecticides may be needed. However, additional applications are only needed if live aphids are still present. It is necessary that before purchasing the suitable insecticide and using it, all label directions must be read and followed, specially the formulation and dosage because the label is the law; therefore, the product label is the final authority on what crop or areas the product can be applied and at what rate. Before purchasing an insecticide, one should be sure to look on the package for active ingredient and select the product with the proper active ingredient to control the pest. Spray should always be done mid- to late evening for best result and to protect beneficial insects and also to avoid any potential plant damage.

3.5.2.4.1 The Use of Insecticidal Soap or Horticultural Oil

The first choice of spray should be insecticidal soap (potassium salt of long-chain fatty acids, 1–2% soap mixed with water) or horticultural oil as the application of these covers the body of the aphids to close the spiracles that results to its suffocation and death. The fatty acids also penetrate the body wall of the aphids and disrupt the cell membranes due to which the cell contents leak out causing the insect to dehydrate and die (Puritch 1981). These solutions when dry after the spray do not leave residue, and any beneficial insect that arrives thereafter will not be contacted with these soaps or oils and remains alive. However, insecticidal soaps are generally ineffective in controlling aphid populations, e.g. leaf curl plum aphid (*Brachycaudus helichrysi*) or the woolly ash aphid (*Prociphilus fraxinifolii*), which are protected inside distorted foliage or galls. Also, the use of soaps or oils should not be applied on water-stressed plants or when the temperature exceeds 32 °C as these may be phytotoxic to some plants. Common aphid species controlled with these types of oils include the woolly apple aphid (*Eriosoma lanigerum*), green apple aphid (*Aphis pomi*), rosy apple aphid (*Dysaphis plantaginea*), mealy plum aphid (*Hyalopterus pruni*), black cherry aphid (*Myzus cerasi*), etc.

3.5.2.4.2 The Use of Biopesticides

The effective step-up from the soaps and oils is application of biopesticides which are derived from plants (botanicals) and are in use in modern agriculture due to their upper hand over synthetic insecticides as usually they are not toxic to nontarget animals and are easily degradable. The use of pyrethrins, nicotine, azadirachtin, rotenone, etc., is time-honoured insecticides; all are commercially available.

3.5.2.4.2.1 Nicotine

Commercially, nicotine (Black Leaf 40) is a formulation of nicotine alkaloid (95%) and nicotine sulphate (40%). The alkaloid is derived from tobacco (*Nicotiana tabacum* and *N. rustica*). It is highly toxic to a great number of insects including aphids as a nerve poison. Since it is also very toxic to humans, it is banned in India but is manufactured for export only. However, the household aqueous preparation of tobacco leaves, garlic and neem gives promising result against cowpea aphid (Bahar et al. 2007).

3.5.2.4.2.2 Pyrethrum

The pyrethrum is extracted from the flowers of *Chrysanthemum* spp. and *Tanacetum cinerariifolium*. It is composed of four compounds: pyrethrins I and II and cinerins I and II. The cinerins are more stable than the pyrethrins. This biopesticide can be very effective in providing a relatively quick knockdown of aphids. This chemical attacks the insects' peripheral nervous system and for this reason has a rapid knockdown; however, the insects soon recover to full activity. Therefore, some synergists are added in the formulation. It is available as spray and dust for use against aphids on fruit trees, vegetables and flowers. Commercial 'Pyrethrum FS' is based on pyrethrin (sesame oil is added as a synergist) against aphids and other sucking insects. The active ingredients are rapidly broken down by sunlight and are only effective for a short time. It has been used against aphids in tomato (Verghese 2015).

3.5.2.4.2.3 Rotenone

Rotenone is found in the roots of several species of plants in the genera *Derris, Millettia*, *Tephrosia* and *Lonchocarpus*. It is probably the second most used botanical. Rotenone is a white to yellowish white crystal and is readily detoxified by the action of air and light. It is a metabolic inhibitor (i.e. inhibits the respiratory chain, the oxidation of NADH-linked substrate) and is a broad-spectrum contact and stomach poison that affects insect nerve and muscle cells, causing the insects to stop feeding and die anywhere from a few hours to a few days after ingestion. Rotenone has been shown to be an effective control agent of many pest species, including aphids (Isman et al. 2011). Yi et al. (2012) have observed effective control of *Aphis gossypii* by the spray of a mixture of rotenone and destruxins. The insecticidal activities of destruxin have been observed against aphids (Robert and Riba 1989).

3.5.2.4.2.4 *Ryania*

Ryania is extracted from the stem and roots of a woody South American plant *Ryania speciosa.* The active ingredient is an alkaloid ryanodine. *Ryania* is a stomach poison that causes insects to stop feeding soon after ingestion. It is reported to be most effective when used in hot weather. *Ryania* has been suggested to use against aphids on trees (Veena 2009).

3.5.2.4.2.5 Azadirachtin

Azadirachtin is the most active compound found in neem (*Azadirachta indica*) plants and is highly toxic to several insect pests such as cotton aphids, cotton bollworms, brown plant hopper, cabbage butterfly, etc. Indeed, the neem plants contain thousands of chemical constituents. Of special interest are the terpenoids that are unique to neem. More than a hundred terpenoids are known from different parts of the neem plant. Azadirachtin is one of the terpenoids. Several different kinds of azadirachtins (A to K) have been isolated, the most abundant of which is azadirachtin A. The neem terpenoids are present in all parts of the plant, in the living tissues especially in the seed kernels. The commercial products of neem (Neem®, Nimbicide®, Achook®, BioNeem®, Neemix®, Azatin®, etc.) work on the metamorphosis of insects. Neem has been used with variable results to manage aphids and other insects. The extract of neem reduces the population of several aphid species causing high mortality and decreasing fecundity, as well as inhibiting population growth in many crops, e.g. *Acyrthosiphon pisum* on pea (Stark and Rangus 1994), *Elatobium abietinum* on spruce (Partridge and Borden 1997), *Aphis craccivora* on cowpea (Ulrichs et al. 2001), *Aphis (Toxoptera) citricidus* on citrus (Tang et al. 2002), *Myzus persicae* on pepper (Shannag et al. 2014), *Sitobion avenae* on wheat (Shah et al. 2017; Matharu and Tanwa 2019) and other aphid pests.

3.5.2.4.3 The Use of Synthetic Insecticides

Both contact and systemic insecticides are available for the control of aphids, primarily on ornamentals, although there are formulations for cereals, legumes, vegetables and fruits. If an insecticide is to be applied on vegetables or fruits, the label on the container must be seen as it will give specific directions as to when the product can be applied prior to harvest. There are hundreds of chemical insecticides with many formulations, but when it is established that chemical control is necessary, a suitable insecticide should be selected. Such insecticides should have following characteristics: (i) it should be safe to nontarget organisms but be highly efficient to kill the target insects; (ii) it should not be phytotoxic nor should it impair the germination of seeds and cause damage to flowers and fruits; (iii) it should not impart off-flavour of food materials; (iv) it should kill the target insects very quickly; (v) it should be persistence in toxicity, i.e. it should maintain lethal action for a longer period; (vi) it should be quickly degradable if persistence is not required; (vii) it should be stable during longer storage; (viii) it should be cheaper and within the reach of poor farmers; etc. (Dewar and Denholm 2017). However, these attributes differ in different situations, and no one insecticide possesses all these desirable attributes. In addition, one should be very careful in applying insecticides in agricultural crops to control aphids.

It should be taken into account that not all insecticides can wipe out aphids. Often the success of any insecticide depends on the timing and way of its application rather than its chemistry. However, different kinds of insecticides are needed to work well for different crops and different aphid species. It has been observed that sometimes combinations of conventional insecticides, such as orthene, endosulfan, metasystox-R, dimethoate and pyrethroids, may be superior. The use of carbaryl against aphids must be avoided as it can be much more detrimental to their natural enemies. Any insecticide should be applied only after proper monitoring the incidence of aphids and its level of infestation exceeding ETL. Table 3.5 displays the type of active ingredients of representative insecticides used nowadays, its formulation, mode of action and possible danger.

Most of the insecticides used to control aphid population are systemic in nature. Chemical control measures for following major agricultural aphid pests are given below.

3.5.2.4.3.1 *Aphis gossypii*

In case of *Aphis gossypii* on cotton, the following steps should be applied to control its population. Seed treatment with imidacloprid 60 FS @ 10 ml/kg seed or with thiamethoxam 70 WS @ 5 g/kg seed keeps the crop free from aphids for a month. If the plant is infested with aphids, spray of following insecticides controls the aphid population: NSKE 5% @ 2.0 ml/l, clothianidin 50 WDG @ 0.075 g/l, imidacloprid 17.8 SL @ 0.25 ml /l, acetamiprid 20 WP @ 0.2 g/l and thiamethoxam 25 WP @ 0.2 g/l.

3.5.2.4.3.2 *Lipaphis erysimi*

The most effective treatments against *Lipaphis erysimi* infesting a seed crop of radish, menazon at 0.17 and 0.34 kg a.i./ha and oxydemeton-methyl at 0.34 kg a. i./ha were recommended, which reduced the aphid population (Sekhon et al. 1980). On cabbage, the treatment of thiamethoxam 0.01%, imidacloprid 0.01%, acetamiprid 0.004% and methyl-o-demeton 0.025% provides considerable control of *Lipaphis erysimi* (Vermora et al. 2010). On cauliflower, it can be controlled by applying cypermethrin 10EC 400 ml/ha and chlorpyriphos 20EC 1000 ml/ha (Krishna et al. 2009) or imidacloprid 17.8 SL @ 0.2 g/l and fipronil 5 SC @ 1.0 ml/l (Dotasara et al. 2017).

The spray of flonicamid 0.02% at seedling stage, flubendamide 0.014% at pre-flowering stage, azadirachtin 0.15% at 50% flowering stage and acephate + fenvalerate 0.028% at 50% pod formation stage keeps the population of *Lipaphis*

Table 3.5 Representative insecticides for aphid control and their active ingredients and mode of action

Type of active ingredient	Representative chemicals	Mode of action	Crops
Anthranilic diamides	Chlorantraniliprole, cyantraniliprolel, flubendiamide	Neurotoxic	Apple, brassica leafy vegetables, cereals, citrus, cotton, cranberries, cucurbit vegetables, grapefruit, grapes, ornamentals, potatoes, rice, spinach, strawberries, sweet potatoes, tomatoes, etc.
Carbamate	Ethiofencarb, pirimicarb, carbaryl	Neurotoxic	Brussels sprouts, cauliflower, celery, cereals, cotton, cottonseed, cranberries, head lettuce, lima beans, macadamia nuts, peanuts, peppermint, peppers, soybeans, spearmint, tobacco, vegetables, etc.
Juvenile hormone analogue	(S)-kinoprene, methoprene	Insect growth regulators	Cereals, vegetables, brassica crops
	Flufenoxuron	Chitin synthesis inhibitor	Brassica crops, tomatoes, potatoes, chillies
Ketoenols	Spiromesifen, spirotetramat	Lipid synthesis inhibitor	Cotton, field corn, chilies, brinjal, apple, tomato, ornamentals, strawberries
Neonicotinoid	Thiamethoxam, acetamiprid, clothianidin, dinotefuran, imidacloprid, thiacloprid, sulfoxaflor, flupyradifurone	Neurotoxic	Apple, leafy vegetables, brassica leafy vegetables, cucurbit vegetables, cotton, cereals, grapes, potatoes, rice, pulses, chillies, ornamentals, etc.
Organochlorine	Endosulfan	Neurotoxic	Soft fruits, vegetables
Organophosphate	Acephate, chlorpyriphos, dichlorvos, dicrotophos, dimethoate, malathion, menazon, methamidophos, methyl demeton, monocrotophos, parathion, phosphamidon, quinalphos	Neurotoxic	Apples, brinjal, brussels sprouts, cauliflower, celery, cereals, cotton, cottonseed, cranberries, chrysanthemums, head lettuce, lima beans, macadamia nuts, pea, peaches, peanuts, peppermint, peppers, potatoes, sugar beet, sweat potatoes, soybeans, spearmint, tobacco, etc.
Phenylpyrazole	Fipronil	GABA agonist	Rice, cereals, cotton, sugarcane, chilli, oilseed rape, vegetables

(continued)

Table 3.5 (continued)

Type of active ingredient	Representative chemicals	Mode of action	Crops
Pyrethroids	Fenvalerate	Neurotoxic	Cotton, corn, soybeans
Pyrethroids	Allethrin, bifenthrin, cyfluthrin, cyhalothrin, cypermethrin, esfenvalerate, fenpropathrin, permethrin, phenothrin, prallethrin, tau-fluvalinate, tetramethrin	Nerve poison	Almost all crops
Pyridine organic compound	Flonicamid	Inhibition of the stylet penetration	Soft fruits, brassica crops, vegetables
Spinosyns	Spinosad, spinetoram	Nerve poison	All crops

erysimi on mustard crop (Kalasariya 2016). Shukla and Mishra (2010) observed promising control of *Lipaphis erysimi* on taramira by applying dimethoate 0.03%, monocrotophos 0.05%, methyl demeton 0.03% and acephate 0.02% proved to be highly effective followed by dimethoate 0.02%, acephate 0.01% and methyl.

3.5.2.4.3.3 *Myzus persicae*

On cauliflower, *Myzus persicae* can be controlled by applying cypermethrin 10EC 400 ml/ha, flufenoxuron (400 ml/ha) and chlorpyriphos 20EC 1000 ml/ha (Krishna et al. 2008). The following insecticides also give satisfactory results when applied in proper concentration and formulation: chinimix, dichlorvos, endosulfan, M.I.P.C., malathion, methyl-o-demeton, phosphamidon, quinalphos, bifenthrin, cyfluthrin, dicrotophos, ethiofencarb, fenvalerate, furadan, methamidophos, parathion, permethrin and pirimicarb (Singh 2015).

3.5.2.4.3.4 *Aphis craccivora*

In case of *Aphis craccivora* on cowpea, the following insecticides give promising result: spray of methyl demeton 25 EC 500 ml/ha or dimethoate 30 EC 500 ml/ha. Three sprays of chlorpyriphos 50% EC and cypermethrin 5% EC @ 2 ml/l after 15 days interval yield considerable control of the aphid (Dhakal et al. 2019).

3.5.2.4.3.5 Cereal Aphids

Worldwide, six species of aphids, viz. bird cherry oat aphid (*Rhopalosiphum padi*), corn aphid (*Rhopalosiphum maidis*), English grain aphid (*Sitobion avenae*) and Indian grain aphid (*Sitobion miscanthi*), attack several cereal crops, such as barley, corn, millets, sorghum and wheat in India. The aphid damage is seen during grain filling stage when both nymphs and adults damage the crops by sucking cell sap from leaves and maturing grains. The infested leaves turn pale, wilt and have a

Table 3.6 Registered insecticides for control of cereal aphids

Active ingredient	Rate per hectare	Remarks
Seed treatment		
Imidacloprid (600 g/l)	120–240 ml/100 kg seed	Higher rate for increased length of control in high-risk areas
Thiamethoxam (210 g/l) + lambda-cyhalothrin (37.5 g/l)	165–330 l/100 kg	Higher rate in areas where higher pest pressure is expected or longer period of control required
Foliar spray		
Acetamiprid 20SP	20 g a.i.	Apply at heading stage
Alpha-cypermethrin (100 g/l)	125 ml	Apply at 3 and 7 weeks after emergence
Beta-cyfluthrin (25 g/l)	250–500 ml	Use high rate in high-risk areas; apply at 5 and 9 weeks post seeding
Chlorantranilipride 18.5 SC	20 g a.i.	Apply at heading stage
Chlorpyriphos (300 g/l)	200–300 ml	Use higher rates if threshold is exceeded
Clothianidin 50 WDG	15 g a.i.	Apply at heading stage
Dimethoate (400 g/l)	500 ml	Apply 7 weeks after
Esfenvalerate (50 g/l)	100–300 ml	Use high rate in high-risk areas; apply after 3 and 7 weeks
Gamma-cyhalothrin (150 g/l)	10–15 ml	Apply sprays 4 and 8 weeks after emergence
Imidacloprid 17.8 SL	20 g a.i.	Apply at heading stage
Lambda-cyhalothrin (250 g/l)	12–18 ml	Apply sprays 4 and 8 weeks after emergence
L-cyhalothrin (15.4 g/l)	200–300 ml	Apply sprays 4 and 8 weeks after emergence
Pirimicarb (500 g/kg)	250–300 g	Apply if aphids have reached threshold
Sulfoxaflor	50–100 ml	Do not apply to crop later than the flag leaf stage
Thiamethoxam 25WG	12.5 g a.i.	Apply at heading stage

stunted appearance and cause heavy grain yield loss. A summary of registered insecticides for use in cereal crops for controlling these aphids is given in Table 3.6.

3.6 Conclusions

The aphids as a group of sucking insects are very fascinating with regard to their morphology, life cycle, behaviour, host–plant interaction, etc. Out of about 5110 species described, about 250 species are considered as notorious pests of hundreds of agricultural and horticultural crops. Some aphids are monophagous, while few are highly polyphagous feeding on hundreds of host–plants. Aphids excrete considerable amount of sugary liquid, honeydew, on which sooty mould usually turns them black that hampers photosynthesis and respiration. The honeydew also serves as food for ants, bees and their parasitic wasps. The aphids are unique on the account of their peculiar mode of reproduction, development and the polymorphism. They may reproduce either by parthenogenesis or zygogenesis. They may either be oviparous or viviparous. Parthenogenetic reproduction allows rapid increase in numbers and results in populations consisting of clones. Some species reproduce both parthenogenetically and sexually (holocyclic species), whereas only few reproduce parthenogenetically (anholocyclic species). The aphids are polymorphic, and both winged (alate) and wingless (aptera) morphs may be found in the same colony. Several factors, both biotic and abiotic, have effect on the formation of different phenotypes, and each phenotype performs different ecological roles in the life history. Aphids are frequently engaged in mutualistic associations with bacterial endosymbionts that not only provide essential amino acids to them but also grant them protection from natural enemies, protection from extreme temperatures, development of resistance to a fungal pathogen and the ability to use a greater diversity of resources.

About 250 species of aphids are major agricultural and horticultural pests of several crops. They damage the crops directly by sucking their nutrients, making galls and hampering photosynthesis and respiration by the growth of sooty moulds on the honeydew deposited thereon. Aphids also damage the crop indirectly by transmitting hundreds of plant viruses. Because of their economic importance, their population must be controlled to save the crops. There are several ways by which the aphids are controlled both in glass houses and fields.

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Whiteflies

4

Shahab Manzari and Yaghoub Fathipour

Contents

Abstract

The family Aleyrodidae is composed of tiny insects, which are usually called whiteflies. The Aleyrodidae includes many sucking species that attack cultivated plants, and several species, e.g., *Bemisia tabaci* (Gennadius), *Trialeurodes vaporariorum* (Westwood), and *Dialeurodes citri* (Ashmead), are economically important as pests of both crops and of ornamental plants. All life stages of whiteflies, except the egg, secrete waxes that cover the body, and this is one of the distinguishing features of the family. The taxonomy of the Aleyrodidae has long been problematic, partly because morphological traits of adults are currently poorly understood and so do not readily permit differentiation between genera or species. Due to their secreted powdery wax, the adults resemble tiny moths

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S. Manzari
Insect Taxonomy Research Department, Iranian Research Institute of Plant Protection, Agricultural Research, Education and Extension Organization (AREEO), Tehran, Iran

Y. Fathipour (✉)
Department of Entomology, Faculty of Agriculture, Tarbiat Modares University, Tehran, Iran
e-mail: fathi@modares.ac.ir

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with which they could, conceivably, be confused. For many years, farmers have been spending a lot of money to control different species of whiteflies in both indoor (greenhouses) and outdoor (fields) cropping systems. In this chapter, different control measures of whiteflies including cultural, biological, and chemical control as well as the use of multiple control methods in a framework of an integrated pest management (IPM) program are discussed in detail.

Keywords

Aleyrodidae · Whitefly · Crop pest · *Bemisia* · *Trialeurodes* · Greenhouse

4.1 Introduction

The family Aleyrodidae is composed of tiny insects (1–3 mm in body length), which are usually called whiteflies, although, rather surprisingly, one of the most widespread pest species, *Aleurocanthus woglumi* Ashby, is called the citrus blackfly (Mound 1973). The common name, "whiteflies," is derived from the white appearance of the powdery wax, which covers the body and wings of the adults of almost all species, and the family name, Aleyrodidae, is derived from the Greek word, *Aàleuron* (*Aleuron*), meaning flour or meal. Mound and Halsey (1978) catalogued the whiteflies of the world and listed 1156 species, which has now increased to approximately 1560 based on the updated checklist of the world's extant whitefly species (Martin and Mound 2007). However, the real number, i.e., including undescribed species, is certainly much higher.

Whiteflies are known from Afrotropical, Australian, Australo-Oriental, Malagasy/Mascarene, Nearctic, Neotropical, Oriental, Pacific, and Palaearctic zoogeographical regions (Mound 1984). Most species are tropical, and even in Europe, the number of species is closely related to latitude (Bink-Moenen and Mound 1990). Three of the most speciose whitefly genera, *Aleuroplatus* Quaintance & Baker, *Aleurotrachelus* Quaintance & Baker and *Tetraleurodes* Cockerell, are known from many parts of the world, but many others have more restricted distributions (Mound and Halsey 1978).

Morphologically and biologically, adult whiteflies resemble Psylloidea, whereas the immature stages are more like those of Coccoidea (Carver et al. 1991). Unusually, among insects, the generic classification of whiteflies is largely based on one of the immature stages, the so-called pupal case (= puparium = fourth instar = last nymphal stage = last larval stage), rather than on the adults (Gill 1990). Quite often, third instar larvae have been used in species descriptions, having been mistaken for the so-called puparial stage (Martin 1999). Unfortunately, whitefly puparia are notorious for displaying variation induced by environmental and physical factors, such as temperature and humidity (Mohanty and Basu 1986), as well as by the type of leaf surface upon which they have developed (Russell 1948; Mound 1963; David and Ananthakrishnan 1976; Neal Jr and Bentz 1999; Guershon and Gerling 2001a). Aleyrodid systematists need to be alert to all of these problems.

4.2 Host Plants

Most whiteflies are known only from angiosperm hosts, primarily dicotyledons. However, some species feed on members of several families of monocotyledons, e.g., Poaceae, Arecaceae, and Smilacaceae. The only record of an aleyrodid species from a gymnosperm is *T. vaporariorum* on *Dioon spinulosum* Dyer (Zamiaceae) (Russell 1963; Mound and Halsey 1978; Dubey and Ko 2008; Evans 2008). In addition to Spermatophyta (including gymnosperms and angiosperms), 20 described, and three or four undescribed, species are known from fern hosts (Pteridophyta) (Mound and Halsey 1978; Martin and Camus 2001).

Few whitefly species are known to be monophagous, most being oligo- or polyphagous. Host specificity does not seem to be highly developed in Aleyrodidae. Worldwide, *Ficus* (Moraceae) is host to more than 80 species of whiteflies and *Citrus* (Rutaceae) to more than 60 (Carver et al. 1991). Well-known polyphagous species, *T. vaporariorum* and *B. tabaci*, have been recorded from more than 120 and 70 plant families, respectively (Mound and Halsey 1978; Greathead 1986; CABI 2019b). Polyphagous species have a range of fitness on different hosts.

4.3 Origin

The paraphyletic nature of Homoptera (including Sternorrhyncha and Auchenorrhyncha) is now widely acknowledged. Efforts to determine the phylogenetic origin of whiteflies have been impeded by gaps in the fossil record of Hemiptera in general and a paucity of fossil whiteflies in particular (Campbell et al. 1994). Because of their size and fragility, adult aleyrodids would not be expected to be preserved as impression fossils often, and still less, their larvae (Evans 1963). The additional possibility that they arose in tropical latitudes may also account for their absence in fossil deposits now located in northern latitudes, where fossils of aphids are found (Shcherbakov and Wegierek 1991). The oldest known fossil whiteflies, *Juleyrodes gilli* Shcherbakov and *J. visnyai* Shcherbakov, belong to the extinct subfamily Bernaeinae and are from the Late Jurassic or Early Cretaceous (more than 140 million years ago) (Shcherbakov 2000).

Based on the phylogenetic analysis of 18S rDNA nucleotide sequences of hemipteran exemplars, the Sternorrhyncha appears to be a monophyletic clade, which forms a sister group to all other hemipterans (the Euhemiptera, including Auchenorrhyncha and Heteroptera) (Campbell et al. 1994, 1995a, 1995b; Von Dohlen and Moran 1995). These molecular findings support several morphologically based cladistic inferences about paraphyly of Homoptera (e.g., Kristensen 1975; Hennig 1981; Popov 1981; Wootton and Betts 1986; Zrzavy 1992). However, relationships within the Sternorrhyncha (which includes psyllids, whiteflies, aphids and scales) are controversial with molecular and morphological data supporting different scenarios. Based on the same molecular evidence, whiteflies form a sister group to aphids and scales, while psyllids form a sister group to all other Sternorrhyncha (Campbell et al. 1994, 1995a). However, some morphological evidence supports a sister-group relationship between whiteflies and psyllids (e.g., Quaintance and Baker 1913; Goodchild 1966; Szelegiewicz 1971; Hennig 1981; Carver et al. 1991; Shcherbakov 2000; Shcherbakov and Popov 2002), though Evans' (1963) morphologically based conclusions are more in agreement with the available molecular studies. Furthermore, some molecular studies focused on the endosymbionts of sternorrhynchans mainly concur with the morphological evidence. The analysis of 16S rDNA sequences of primary bacterial endosymbionts supports a sister relationship between primary endosymbionts of psyllids and whiteflies (Spaulding and von Dohlen 1998, 2001).

The molecular genetics of insect endosymbionts suggests that several independent lineages of intracellular bacteria have followed similar paths of evolution, and for several insect taxa, there is a pattern of congruent host and primary bacterial endosymbiont phylogenies (Moran and Telang 1998). These support the idea that extant associations arose from a single infection of the common host ancestor followed by vertical transmission of symbionts and parallel diversification of insect and bacterial lineages (Munson et al. 1991; Moran et al. 1993; Moran and Telang 1998). In the same way, because most extant sternorrhynchans feed on phloem sap, it is plausible to hypothesize that their common ancestor was a phloem feeder and had endosymbionts to supplement its diet (Spaulding and von Dohlen 1998).

4.4 Life History

Information on basic biology is scarce for most species and mainly restricted to a few well-known pest species such as *B. tabaci* and *T. vaporariorum* (Byrne and Bellows 1991). Whiteflies have six life stages, the egg, the crawler (first nymphal instar or stadium), three sessile nymphal instars (second, third, and fourth instar nymphs, the latter being termed "pupa"), and the adult or imago (Gill 1990) (Fig. 4.1).

The eggs are pedunculate like those of Psylloidea, usually elongate-oval in shape but occasionally reniform, and are either smooth or sculptured. The pedicel, a hollow extension of the chorion, is either inserted into a stomatal opening or into a slit made by the ovipositor in the leaf surface (Quaintance and Baker 1913; Gill 1990; Carver et al. 1991). Two main functions have been reported for the pedicel in addition to providing a means of attachment. According to Quaintance and Baker (1913), the pedicel seems to direct the spermatozoa at the time of fertilization, during which time it is filled with protoplasm. These authors also stated, "After fertilization, the protoplasm dries up and the pedicel becomes a hollow tube." Other authors have suggested that the pedicel is used as a means of absorbing water into the egg, which protects the egg from dehydration (Poinar 1965; Paulson and Beardsley 1985; Byrne et al. 1990b). Based on the study (Buckner et al. 2002) on *B. tabaci*, biotype B (previously known as *Bemisia argentifolii* Bellows & Perring) using scanning and transmission electron microscopy, the exterior of the distal portion of the pedicel comprises a tangled array of fibrous structures (0.2–0.3 mm in diameter) that constitutes about 20–25% of the outer diameter of the pedicel. The attachments of the fibers to the core of the pedicle suggest that the pedicel may function as a

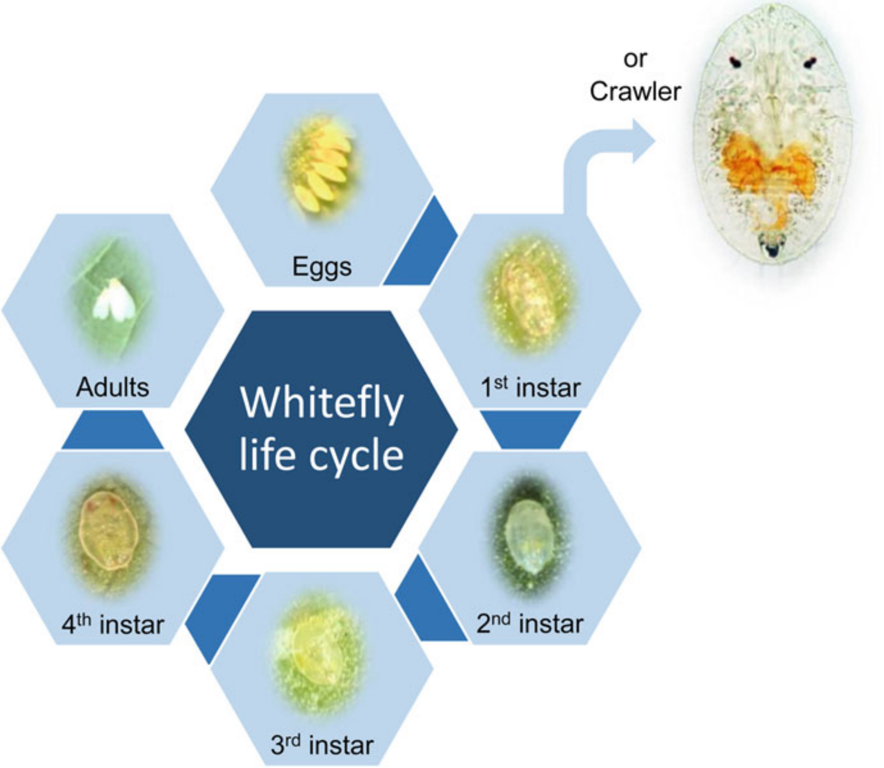

Fig. 4.1 Life cycle of whiteflies

collector and conduit for water (vapor) and perhaps solute movement into the egg (Buckner et al. 2002). These authors also studied the effect of exposure of pedicels to different relative humidities and to radiolabeled materials and demonstrated that whitefly egg hatch is dependent on water uptake by the pedicel and that the pedicel also has the ability to transport solutes into the developing egg.

The crawler has functional walking legs (apparently three-segmented) and antennae (apparently two-segmented) (Fig. 4.1, inset). It is the only mobile immature stage. In most species, the crawler probably settles within a few millimeters of the egg. Once crawlers find the phloem of a host plant, they remain immobile (with the exception of short periods during molts) until they become adults (Gill 1990; Byrne and Bellows 1991).

The second and third instar nymphs are intermediate feeding stages and are usually oval or oval-elongate and similar in general shape and color to the pupal case. The two subfamilies of the Aleyrodidae, Aleyrodinae and Aleurodicinae, differ morphologically in these stages. Legs and antennae appear to be reduced to one segment in the Aleyrodinae, but in the Aleurodicinae the legs are three-segmented and the antennae are without distinct segmentation (Gill 1990). These stages are not known for the third subfamily, Udamoselinae (Martin 2007).

The fourth instar is not a true pupa as seen in holometabolous insects. Based on studies on *T. vaporariorum*, it has three morphologically distinct forms (Nechols and Tauber 1977a, b). The early fourth instar is flattened and translucent, but unlike the true pupa, it continues to feed. The next form, termed the transitional substage, is expanded and opaque white with dorsal and lateral waxy, spine-like processes. The separation of the old cuticle from the underlying epidermal cells (apolysis) takes place at this point. In the last form, the pharate adult with red eyes and yellow body pigment is present. This is the stage that apolysis is complete and the adult cuticle is laid down (Nechols and Tauber 1977a, b). Dividing the fourth instar into only three stages would prevent the detection of important physiological/biochemical changes that occur during the development of the fourth instar and the pharate adult (Gelman et al. 2002), and these authors divided the fourth instar *T. vaporariorum* into nine stages: the last two of the above three forms were further subdivided, and they provide additional detail of morphological changes of the eye and wing as well as changes in soluble protein content and whole-body ecdysteroid fluctuations.

In most species, the adult emerges through a T-shaped split in the dorsal surface of the pupal case, but in few species the apices of the T are connected by additional sutures, thus forming a “trapdoor” (Mound and Halsey 1978). Males and females are similar in appearance except for their genitalia; however, male whiteflies are often smaller than females of the same species.

All life stages of whiteflies, except the egg, secrete waxes that cover the body (Byrne and Bellows 1991), and this is one of the distinguishing features of the family. Waxes can be secreted as both waxy particles and filaments. In adults, the ventral wax plates seem to be somewhat unique in each subfamily. In the Aleyrodinae, there are two pairs of wax plates on the second and third abdominal segments of females and four pairs on the second to fifth abdominal segments of males (Gill 1990). However, the aleyrodine genus *Neomaskellia* Quaintance & Baker has two reduced wax plates in males (Bink-Moenen and Mound 1990). In the Aleurodicinae, there are four pairs of wax plates in females on abdominal segments II–V and three pairs in males on segments II–IV (Gill 1990). Females are unknown for the Udamoselinae, but the abdominal segments III–V in males each bear a pair of wax plates (Martin 2007).

The external lipids of the adult whitefly can be differentiated into two categories: the wax particles and the lipids of the cuticular surface (Nelson et al. 1997). These authors’ studies on selected species of whiteflies show that the wax particles are not a true wax but are composed of long-chain aldehydes and long-chain alcohols. However, the lipids of the cuticular surface are composed of true wax esters and small amounts of hydrocarbons (Nelson et al. 1997). The main function of cuticular lipids on terrestrial insects is to minimize loss of water across the integument. Other widely accepted functions include defense mechanisms against predators, chemical communication, and camouflage. During growth and development, whitefly nymphs remain immobile and feed continually on plant phloem except during molting activities between instars. Therefore, a need for cuticular lipids in reducing water loss could be a less important function than the role of cuticular lipids in protecting sessile nymphs against predators and providing camouflage. For whitefly nymphs, the waxy coating on the cuticular surfaces could also function to protect against honeydew, the sticky liquid excreted by phloem feeders (Buckner et al. 1999).

Reproduction in whiteflies is usually sexual, but occasionally it is parthenogenetic (Martin et al. 2000). Unmated females, except in *Parabemisia myricae (Kuwana),* , produce male offspring (XO), and mated females may produce both males and females (XO and XX) (Byrne and Bellows 1991). Two races of *T. vaporariorum*, an American and an English one, show arrhenotokous and thelytokous parthenogenesis, respectively (Schrader 1920). According to Ahman and Ekbom (1981), however, virgin females did not give rise to females in any *T. vaporariorum* cultures studied by them. Females usually deposit their eggs on the lower surfaces of leaves, and the eggs of many species are laid in partial or complete circles by rotation of the female around her rostrum. Some species, particularly in the Aleurodicinae, oviposit on other surfaces such as fruits, and a few whitefly species habitually develop on the upper surfaces of leaves, while others readily develop on both surfaces (Martin et al. 2000).

Adult males of a few whitefly species, especially *B. tabaci* and *T. vaporariorum*, have been observed to display courtship behavior prior to mating, including abdominal oscillations that result in acoustic signals (substrate-borne vibrations) (Li et al. 1989; Kanmiya 1996; Kanmiya and Sonobe 2002; Gullan and Martin 2003). Females may produce sex pheromones to attract males (Li and Maschwitz 1983). Adult whiteflies fly short distances if disturbed from their host plant. They also undertake longer migratory flights, which are air-current dependent because whiteflies are weak fliers (Gullan and Martin 2003). However, based on Byrne's (1999) study on *B. tabaci*, whitefly flight may not be characterized as weak. Whiteflies flew in a flight chamber against a strong air stream as well as dispersed in field experiments for a considerable distance. There is also information that whiteflies have a migratory form, as some strong flying insects, and their flight seems to meet many criteria associated with migration in insects (Byrne 1999). Whiteflies have complex host-finding and host-orientation behavior, involving at least attraction to particular colors, especially yellow or yellow green (Gullan and Martin 2003).

Parental care in whiteflies has been reported in *Aleyrodes singularis* Danzig (Guershon and Gerling 1994, 2001b) and *Neomaskellia bergii* (Signoret) (Kurosu et al. 1992). The findings of Guershon and Gerling (2001b) showed that specific behavioral displays, e.g., "bout flutter," "wing hit," and "rodeo," performed by adult *A. singularis* under laboratory conditions, in the presence of their natural enemies, may cause the latter to depart from the patch and so enhance the survival chances of the immature stages. Therefore, in *A. singularis*, parental care exists at a level considered to be beyond passive protection of eggs or even nymphs (Guershon and Gerling 2001b).

Unlike some aphids that use major veins as feeding sites (Gibson 1972), in at least some whiteflies, *B. tabaci*, biotype B, successful feeding always involves probing of minor veins that contain no more than three xylem elements (Cohen et al. 1998).

Therefore, whitefly feeding biology is complex and includes the location of appropriate sites to probe leaves so that minor veins can be located. There are distinct surface structures such as lamina trichomes and elongated epidermal cells that serve as guides for crawlers to begin probing at appropriate places. The nymphs use a specialized saliva to produce a sleeve-like structure named the "salivary sheath" that guides the stylets to the minor veins. The sheaths are often sinuous and may be branched, both in the mesophyll and at the veins (Cohen et al. 1998).

Whiteflies as well as other insects that feed throughout their life cycles on nutritionally incomplete diets such as plant sap, blood, and wood often harbor mutualistic intracellular prokaryotes that apparently are vital and provide specific nutritional needs for survival and reproduction of the hosts (Douglas 1998; Spaulding and von Dohlen 1998). Such endosymbionts live within specialized host cells called mycetocytes or, more appropriately, bacteriocytes (Spaulding and von Dohlen 1998, 2001), which are organized into a large specialized organ-like structure called the mycetome or bacteriome (Campbell 1993; Spaulding and von Dohlen 1998; Baumann 2005). Some of the prokaryotic symbionts in whiteflies are transient or nonessential that can either be beneficial or deleterious in the long term (Zchori-Fein and Brown 2002). The most distinctive groups of prokaryotic symbionts are "primary" or "obligatory" and "secondary" or "facultative" symbionts (Moran and Baumann 2000; Zchori-Fein and Brown 2002). Primary symbionts are strictly maternally inherited, essential for the host's survival and reproduction, and confined to bacteriocytes (Fukatsu and Nikoh 1998; Moran and Telang 1998; Moran and Baumann 2000; McCutcheon and Moran 2010; Russell et al. 2013). They are believed to represent an ancient mutualism (Spaulding and von Dohlen 1998, 2001). In contrast, secondary symbionts are not necessarily incorporated into bacteriocytes nor are they essential to host survival (Chen and Purcell 1997; Fukatsu and Nikoh 1998; Fukatsu et al. 2000). They are crucial perhaps for fitness, competition, and other more indirect aspects that also influence the evolution of their host (Zchori-Fein and Brown 2002).

4.5 Taxonomy

The taxonomy of the Aleyrodidae has long been problematic, partly because morphological traits of adults are currently poorly understood and so do not readily permit differentiation between genera or species (Frohlich et al. 1999). Due to their secreted powdery wax, the adults resemble tiny moths with which they could, conceivably, be confused. In fact, early workers thought they were minute moths, and *Aleyrodes proletella* (L.) was mistakenly classified in the Lepidoptera by Linnaeus (1758). Later, in 1795, Latreille (1795) correctly recognized their hemipterous nature, and Westwood (1840) established the family name Aleyrodidae. The basis of the generic classification of the family was laid by Quaintance and Baker (1913, 1914).

Whiteflies belong to the order Hemiptera, suborder Sternorrhyncha[1], and superfamily Aleyrodoidea (Martin et al. 2000; Martin 2004); however, based on the classification proposed by Shcherbakov and Popov (2002), they belong to the superorder Cimicidea, order Hemiptera (= Cimicida = Homoptera + Heteroptera), and suborder Psyllina. In the latter classification, Sternorrhyncha splits into two suborders, Psyllina and Aphidina. Aleyrodidae were almost universally considered to belong to Homoptera: Sternorrhyncha until relatively recently when molecular phylogeny showed “Homoptera” unequivocally to be a paraphyletic taxon (Wheeler et al. 1993; Campbell et al. 1994; Campbell et al. 1995b; Sorensen et al. 1995; Von Dohlen and Moran 1995).

The Aleyrodoidea includes the single family, Aleyrodidae, and is supported by the following synapomorphies: “forewing with RS concave, M reduced, both CuA and 1A+2A simple; fore- and hindwing uncoupled in flight; metathorax enlarged; tibiae with setal rows used for wax distribution; abdomen petiolate, desclerotised, with spiracles reduced up to 2 pairs; ovipositor lacking sheath; anal tube transformed into operculum bearing lingula” (Shcherbakov and Popov 2002). The latter structure, which is known as the vasiform orifice, is unique to Aleyrodidae and present in all larval stages, as well as in the adults. Hence, it provides a simple character for recognizing whiteflies (Martin 2004). It is worth mentioning that Maskell (1895) was the first to describe and name the vasiform orifice and its components the lingula, operculum, and lingular setae.

Five extant subfamilies have been established at various times within the Aleyrodidae, of which only three are now considered valid: the Aleurodicinae, found mainly in Central and South America; the Aleyrodinae, which are more widespread (Mound and Halsey 1978; Gill 1990); and the Udamoselinae, suggested on the basis of a single specimen of a South American species, a male with a body length of 7 mm (Enderlein 1909), which are represented today by only two species in a single genus, *Udamoselis* Enderlein (Martin 2007; Martin and Mound 2007). The Aleyrodinae has been divided into 13 tribes: Aleurocanthini, Aleurochitonini, Aleurolobini, Aleuroplatini, Aleyrodini, Bemisini, Dialeurodini, Lipaleyrodini, Neomaskellini, Siphoninini, Tetraleurodini, Trialeurodini, and Zaphanerini (Sampson 1943; Russell 1947; Takahashi 1954; David 1990); however, the applicability of any tribe level classification for the Aleyrodinae seems to be premature due to our current poor understanding of the significance of puparial characters (Manzari and Quicke 2006).

The basic morphological differences between pupae of the Aleyrodinae and Aleurodicinae are the frequent presence of compound/agglomerate pores on the dorsal disk and the presence of a single claw at the apex of each leg in the

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[11] The name “Sternorrhyncha” comes from the Greek *sternon*, meaning chest or breast, and *rhynchos*, meaning nose, snout, or muzzle, and refers to the ventral location of the mouthparts (between the bases of the front legs) of these insects.

Aleurodicinae. In the Aleyrodinae, the former character is absent, and the legs end in a disk (adhesion pad) rather than a claw (Gill 1990; Martin 1999; Russell 2000). Additionally, the number of lingular setae (usually four in the Aleurodicinae and two or none visible in the Aleyrodinae) has been used as a diagnostic character (Gill 1990; Martin et al. 2000). Pupae are unknown for Udamoselinae (Martin 2007).

4.5.1 *Bemisia tabaci* (Gennadius)

Bemisia tabaci was firstly described as *Aleurodes tabaci*, the tobacco whitefly, from Greece by Gennadius (1889), but Takahashi (1936) later transferred it to the genus *Bemisia* Quaintance & Baker. It was subsequently described under several names before its host-correlated morphological variation was recognized. As a result, 20 species of whiteflies were synonymized with *B. tabaci* (Russell 1957; Mound and Halsey 1978; Perring 2001; Martin and Mound 2007). Based on partial mitochondrial cytochrome oxidase subunit I (mtCOI), *B. tabaci* is currently considered as a species complex with at least 45 morphologically indistinguishable, or cryptic, species (including the five recently discovered species from Africa [Mugerwa et al. 2018]) delimited by 3.5% mtCO1 sequence divergence (Dinsdale et al. 2010; Tay et al. 2012; da Barbosa et al. 2014; Boykin and De Barro 2014; Polston et al. 2014; Alemandri et al. 2015; Jiu et al. 2017). However, Lee et al. (2013) proposed a genetic divergence of 4% to discriminate species. Different nomenclatures have consequently been proposed to refer to the entity initially called *A. tabaci*, including biotype, race, genetic group, putative species, species, and haplotype group (Boykin 2014).

Bemisia tabaci is one of the most invasive and devastating pests of both field and protected agricultural crops and ornamental plants in tropical, subtropical, and temperate regions. This is partly due to its adaptability, diverse host plant range, and capacity to vector plant viruses. The species is polyphagous, feeding on an estimated 900 plant species (Mound and Halsey 1978; Greathead 1986; Oliveira et al. 2001; Simmons et al. 2008; Li et al. 2011; McKenzie et al. 2014). Such an extreme host plant range is actually related to all of the known cryptic species that have been allocated to the single taxon, *B. tabaci*. In fact, with a few exceptions, most of the species in the *B. tabaci* complex have a limited host range (Perring et al. 2018). Two species, the Middle East-Asia Minor 1 (MEAM1, formerly B biotype or *Bemisia argentifolii* Bellows & Perring) and Mediterranean (MED, formerly Q biotype) species, which are considered as the most invasive and broadly distributed species around the world (da Barbosa et al. 2014; Jiu et al. 2017; Mugerwa et al. 2018), are highly polyphagous, feeding on many host plants (Wang et al. 2011; Götz and Winter 2016; Perring et al. 2018), but some (e.g., the biotype E and Jatropha race/biotype) are only known from a single host plant and seem to be monophagous (Frohlich et al. 1999; Perring 2001; De Barro et al. 2011).

Diagnostic Features *Egg*. Adult females of *B. tabaci* either scatter eggs or lay them in a partial circle on the underside of leaves; however, egg deposition in the latter form is rare. They are white when first laid but turn a light golden-brown color a few days after being laid. Eggs are oval-shaped and smooth, with a slender and short pedicel at the broader end. Pedicel is about one sixth of the length of egg. When the crawler (first nymphal instar) emerges, the egg chorion remains erect (Hill 1969; Bink-Moenen 1991; Carapia Ruiz and Castillo-Gutiérrez 2013; Perring et al. 2018).

Pupal case. It is usually colorless and suboval in shape. Living pupae are yellowish transparent, flat, and not elevated from the leaf surface; tufts of white wax are often present in the thoracic tracheal pore areas. In slide-mounted specimens, the margin is finely and irregularly crenulated and often slightly curved inwardly at caudal and thoracic tracheal openings. Transverse molting sutures terminate in subdorsum; longitudinal molting suture reach the margin of pupal case. Submargin is with eight or nine pairs of minute setae, three or four pairs in cephalothorax (the fourth pair is usually lacking) and five pairs posterior to fourth abdominal segment (the fifth pair may be well developed); submarginal setae are often hardly discernible; dorsal disk is with five to six pairs of setae (including nominal cephalic, and first and eighth abdominal pairs); these setae are highly variable in size and minute or very well developed. Caudal setae are always stout, usually as long as vasiform orifice or more. Vasiform orifice is triangular, straight-sided (Fig. 4.3-a), and inset from posterior margin of pupal case by less than its own length; operculum covers about anterior half of orifice; lingula is spatulate and finely spinulose, with a pair of terminal setae; vasiform orifice with some posterior tubercles (Fig. 4.3-a, arrow). As with other species of the genus, the length of abdominal segment VII is much reduced medially in *B. tabaci*, so that the abdomen may superficially be appearing seven-segmented (Martin 1987; Bink-Moenen 1983, 1991; Bellows Jr et al. 1994; Martin et al. 2000).

Adult. The body is yellow to dark yellow, and the wings are hyaline. The body and wings are covered with powdery white wax; the wings are held at a "roof-like" angle over the abdomen when at rest. The antennae are seven-segmented. Segment III is the longest, approximately as long as antennal segments IV–VII combined; segments V–VII are subequal in length; segment VI is with a pit sensorium. Upper and lower compound eyes are connected by a single ommatidium; ommatidia in the upper compound eye are smaller than those in lower compound eye. In bleached specimens, they are colorless in upper compound eye but arranged in a pattern of interconnected groups of six pigmented ommatidia surrounding a single nonpigmented ommatidium in lower compound eye (Bink-Moenen 1991; Bellows Jr et al. 1994).

4.5.2 *Trialeurodes vaporariorum* (Westwood)

The "greenhouse whitefly" or "glasshouse whitefly," *Trialeurodes vaporariorum*, was originally described from England as *Aleyrodes vaporariorum* by Westwood in Westwood 1856 and then later transferred by Quaintance and Baker (1915) to the genus *Trialeurodes* Cockerell. The author, Westwood, assumed the species to be a

New World native, being possibly imported to England from Mexico. According to Russell (1948), *T. vaporariorum* is indigenous to western and southwestern part of North America.

The greenhouse whitefly is well known as a worldwide pest of crops in open field and greenhouse situations and, together with *Bemisia tabaci*, is one of the most economically important whitefly species (Martin 1999; Martin et al. 2000; Hernández-Suárez et al. 2012; Perring et al. 2018). It is also a highly polyphagous species and, similar to *B. tabaci*, seems not to rely on host-specific visual or olfactory cues for host finding (Mound 1962; van Lenteren and Noldus 1990). The species is colonizing about 280 host plants from more than 80 families (Mound and Halsey 1978); however, it is currently estimated to exist on 859 species of plants belonging to 469 genera and 121 families (CABI 2019b).

Diagnostic Features *Egg*. Adult females of *T. vaporariorum* generally lay eggs in neat circles or semicircles rather than scatter them on the underside of leaves. Eggs are yellowish white in color when first laid but become dark brown to black within a few days. They are smooth, elongated ovoid in shape, and broadly rounded at the base where a short pedicel is attached. When the crawler emerges, the egg chorion becomes flattened laterally and bent with the apex directed downwards (Hill 1969; Oana et al. 2007; Carapia Ruiz and Castillo-Gutiérrez 2013; Voigt et al. 2019).

Pupal case. It is usually colorless and oval in shape. Living pupae are opaque yellowish white, elevated from the leaf surface, and surrounded by downward curving wax filaments; dorsal glassy wax rods are present. In slide-mounted specimens, the margin is crenate and modified at caudal and thoracic tracheal pores as subtle combs of smaller crenulations. Transverse molting sutures reach the submargin of pupal case; longitudinal molting suture reaches the margin. Submargin is with a single row of well-developed papillae, one to nine (usually four to five) pairs distinctly larger than the others; usually four pairs of such papillae are in subdorsal-submedian position. Cephalic, first and eighth abdominal, and caudal setae are present, variating in length. The vasiform orifice is subtriangular and elongate-cordate (Fig. 4.3-b). The operculum fills about anterior half of orifice. The lingula is lobular, finely spinulose, and with a pair of terminal setae; the apical notch of vasiform orifice is with a minute tongue-like structure, usually protruding beyond the notch. The same as with *B. tabaci*, the length of abdominal segment VII is much reduced medially (Hill 1969; Martin 1987; Bink-Moenen and Gerling 1990; Martin et al. 2000).

Adult. The body is pale yellow. The body and wings are covered with powdery white wax; the wings are held more flatly over the abdomen, nearly parallel to the leaf surface, when at rest. The antennae are similar to those of *B. tabaci* but with no pit sensorium on segment VI. Upper and lower compound eyes are completely separated, not connected by a single ommatidium; the upper compound eye is with ommatidia smaller than those in lower compound eye. Adults are slightly larger than those of *B. tabaci* (Hill 1969; Carapia Ruiz and Castillo-Gutiérrez 2013).

4.6 Nature of Damage

The Aleyrodidae includes many species that attack cultivated plants, and several species, e.g., *B. tabaci*, *T. vaporariorum*, and *Dialeurodes citri* (Ashmead), are economically important as pests of both crops and of ornamental plants. Further, the importance of whiteflies as economic pests seems to be increasing constantly. The amount of damage caused by whiteflies is considerable; for example, in 1991, *B. tabaci* caused damage to the US agricultural production estimated at over 500 million dollars (Perring et al. 1993). These hemipteran insects damage crops by direct feeding on large quantities of phloem sap, which can result in reduced crop yield and plant vigor. With high whitefly populations, plants may die. Furthermore, the honeydew excreted by these insects can serve as a medium for sooty mold fungi (e.g., *Capnodium* spp.) that undesirably affect photosynthesis, which may severely reduce the productivity of the plant (Perkins 1983; Byrne et al. 1990a). In addition, sooty molds may increase thermal absorption and raise leaf temperature, reducing leaf efficiency and possibly even causing premature death of tissue (Byrne et al. 1990a). Finally, only five species of whiteflies, *B. tabaci* species complex (cotton/tobacco/sweet potato whitefly), *T. vaporariorum* (greenhouse whitefly), *Trialeurodes abutiloneus* (Haldeman) (banded-winged whitefly), *Parabemisia myricae* (Kuwana) (Japanese bayberry whitefly), and *Bemisia afer* (Priesner & Hosny) *sensu lato*, can transmit viruses between plants (Kersting et al. 1996; Gamarra et al. 2010; Navas-Castillo et al. 2011; Lapidot et al. 2014). *Bemisia tabaci* species complex is the vector of viruses in several groups, the largest of which is the family Geminiviridae, and it is known to transmit more than 190 species belonging to the genus *Begomovirus* (Lapidot et al. 2014; Polston et al. 2014). It is also considered as one of the world's top 100 invasive species (Lowe et al. 2000). Surprisingly, despite the known importance of whiteflies as virus vectors, the Aleyrodidae is a relatively poorly studied family of insects.

4.7 Management Measures

Whiteflies have long been considered as an important insect pest on a global scale, attacking a wide variety of agricultural crops (Perring et al. 2018). Although there are about 1560 whitefly species in the world (Martin and Mound 2007), only a few species like *B. tabaci*, *T. vaporariorum*, *D. citri*, and *Aleurothrixus floccosus* (Maskell) cause most of the damage worldwide (see also Figs. 4.2 and 4.4). Therefore, in this chapter, we focus only on the management of economically important species, especially *B. tabaci* and *T. vaporariorum*.

4.7.1 Cultural Control

The use of all available environmental and physical tools, as well as spatial and temporal manipulation of pests including the use of traps, resistant cultivars, banker

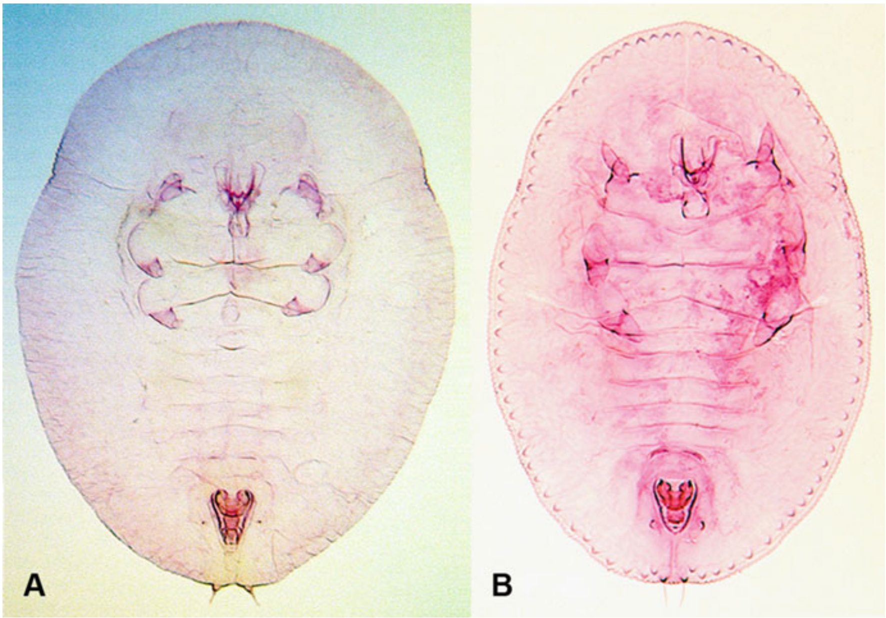

Fig. 4.2 The most common and economically important whitefly species: (**a**) *Bemisia tabaci*, (**b**) *Trialeurodes vaporariorum*

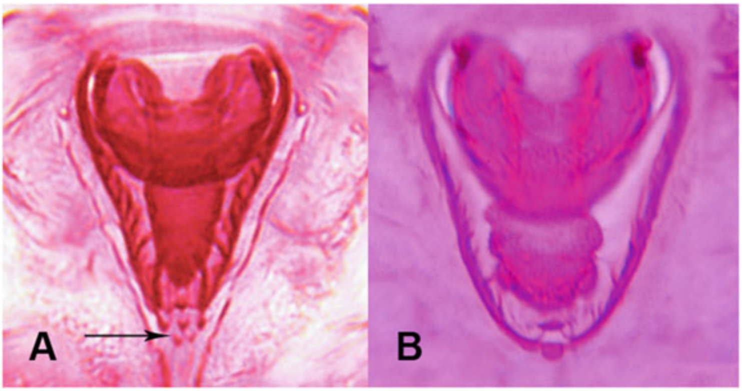

Fig. 4.3 Vasiform orifice: (**a**) *Bemisia tabaci*, (**b**) *Trialeurodes vaporariorum*. Arrow indicates tubercles

and trap plants, reflective and oil-coated yellow mulches, irrigation and fertilizer management, early or delayed planting, manipulation of the type and amount of cultivation, changing the crop type, intercropping, light manipulation, destroying crop residues, and emptying the field, can help pest population management on the field under the heading "cultural control." It is noteworthy that these methods should

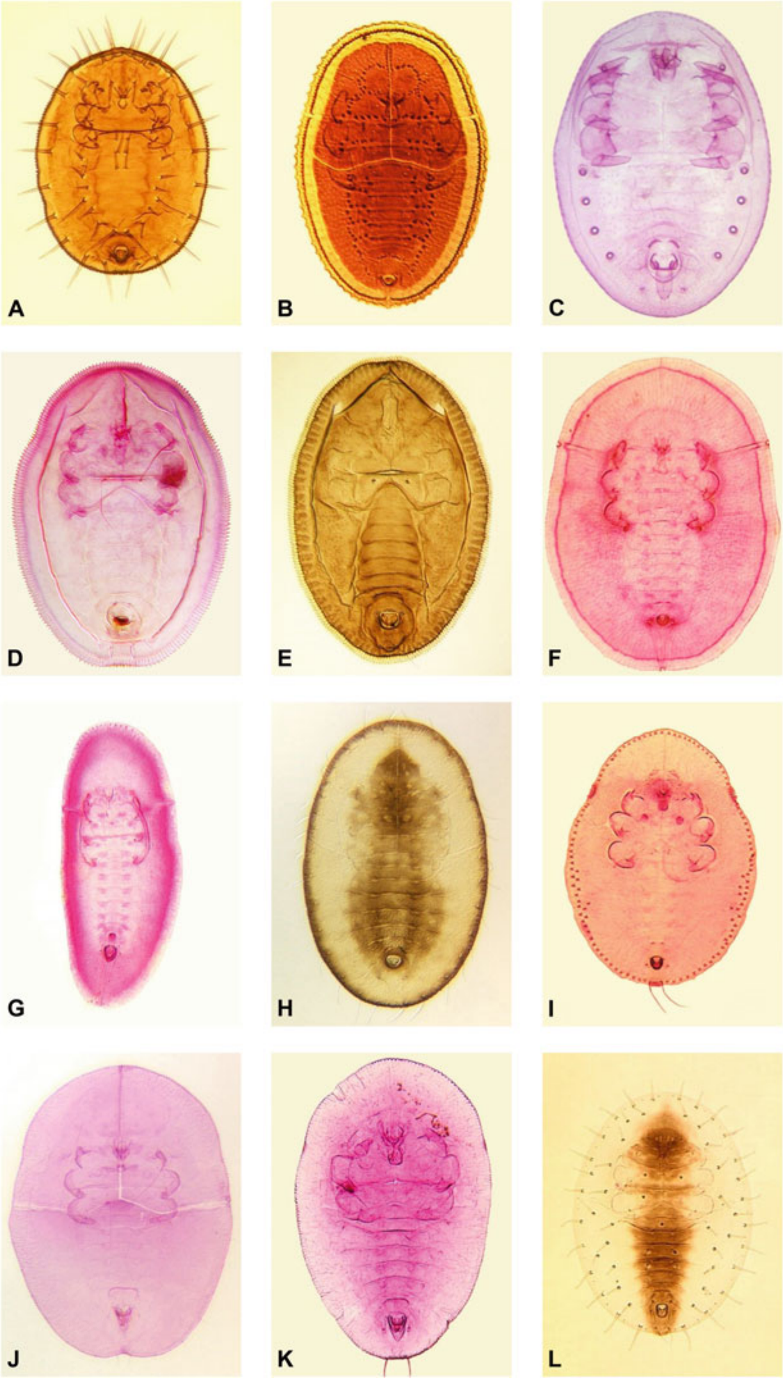

Fig. 4.4 Some common whitefly pest species of the world (see also Fig. 4.2). (**a**) *Aleurocanthus spiniferus* (Quaintance), (**b**) *Aleuroclava neolitseae* (Takahashi), (**c**) *Aleurodicus dispersus* Russell, (**d**) *Aleurothrixus floccosus* (Maskell), (**e**) *Aleurotrachelus trachoides* (Back), (**f**) *Dialeurodes citri*

be carefully used to minimize possible negative impacts on natural enemies released or present in the environment (Wang and Yang 2017). Drought stress and excessive consumption of nitrogen fertilizer improve conditions for whiteflies to increase their populations and the amount of honeydew production (Flint et al. 1996; Bi et al. 2005). Fertilization by vermicompost creates both antixenosis (nonpreference) and antibiosis (decreases survival and prolongs development time) resistance of tomato plants to *B. tabaci*. Pistachio waste vermicompost (PWV) significantly increases preadult development time by 24.65 days and mortality by 54.11%, while these parameters for sterile plant growth media without fertilization are 16.51 days and 7.91%, respectively. The adults show low preference to settling and oviposition on plants treated with vermicomposts and lay significantly fewer eggs in PWV treatment in comparison with the control plant. These results could be due to phenolic compounds in pistachio waste. In addition to pistachio waste that causes suitable growth and resistance to tomato plants, date waste and food waste also have positive effect on plants in resistance to sweet potato whitefly (Sedaghatbaf et al. 2018).

Intercropping prevents accurate identification of the host by the pest and reduces whitefly establishment time on one plant, especially commercial products, and subsequently decreases insect damage by providing a collection of host plants for the whitefly (Bernays 1999). Tomato intercropping with coriander, *Coriandrum sativum* L., followed by sprinkler irrigation reduces appropriateness of the host plant (tomato) for being selected by the whitefly and *Begomovirus* incidence and also the adult and nymph populations of *B. tabaci*. This combination of cultural methods improves the management of the whitefly and its associated virus, *Begomovirus*, which can be due to olfactory and visual obstacles of coriander and physical obstacles of the irrigation method. Therefore, it is an efficient application in organic tomato cultivation systems (Togni et al. 2018). It has also been reported a reduction of whitefly density and virus infection in cucumber, green bean, squash, and tomato plants using a daily drip irrigation method (Abd-Rabou and Simmons 2012).

It has been demonstrated that *Cucumis melo* L. (melon) and *Ricinus communis* L. (castor bean) are suitable banker plants for *B. tabaci* and its parasitoids in tomato cultivation (Kidane et al. 2018). Papaya plant can also be a banker plant for *Encarsia sophia* (Girault & Dodd) as a biocontrol agent of *B. tabaci* MEAM1. This plant is an excellent non-crop banker plant for supporting the non-pest alternative host, *Trialeurodes variabilis* (Quaintance). The latter species is used as an alternative host for rearing of the parasitoid wasp to attack the target pest. The parasitoid travels 14.5 meters between papaya and tomato to reach *B. tabaci*, and there is no significant difference between the percent parasitism on *B. tabaci* (29–45.9%) and *T. variabilis* (36.2–47.4%). Therefore, this system provides a new option for long-term control of *B. tabaci* in greenhouse vegetable production (Xiao et al. 2011). Banker plants also

Fig. 4.4 (continued) (Ashmead), (**g**) *Dialeurolonga elongata* Dozier, (**h**) *Neomaskellia andropogonis* Corbett, (**i**) *Orchamoplatus citri* (Takahashi), (**j**) *Parabemisia myricae* (Kuwana), (**k**) *Pealius azaleae* (Baker & Moles), (**l**) *Siphoninus phillyreae* (Haliday) guarantee continuous presence of biological agents in field cultivation (Parolin et al. 2012).

Phytosanitary irradiation (PI) is a promising and effective method for controlling insect pests such as whiteflies, which can also be applied after harvesting (Hallman 2011). Effective dose of PI does not significantly affect the quality of the fresh products. Another feature of this method is the effectiveness of a treatment dose on a wide range of pests, although different life stages of a pest species require different doses (Hallman et al. 2010). PI does not kill all individuals of a pest population, but it prevents the completion of its developmental stages, reproduction, and oviposition. The generic dose of 100 Gy has been suggested for Aleyrodidae (Hallman 2012), while 108 Gy has also been proposed for *T. vaporariorum* (Van Nieuwenhove et al. 2016). In the recent research, 33,625 late pupae were tested, of which only 1146 (3.4%) emerged, and, interestingly, they did not lay any eggs. Therefore, PI might be a low-risk method for the treatment of sensitive exported commodities.

4.7.2 Biological Control

To date, a lot of research has been carried out worldwide on the use of natural enemies to control the various species of whiteflies. The natural enemies of whiteflies include a long list of parasitoids and predators mainly belonging to Hymenoptera and Coleoptera, as well as some Diptera, Hemiptera, Lepidoptera, Neuroptera, Thysanoptera, and Acari (Arachnida) (Mound and Halsey 1978) (for some examples, see Fig. 4.5). Furthermore, some entomopathogenic fungi have been reported as pathogens of whiteflies (Evans 2008; James 2008). The Centre for Agriculture and Bioscience International (CABI) in its latest update in March 2018 listed 88 and 55 natural enemy species for two of the most damaging whitefly species, *B. tabaci* and *T. vaporariorum*, respectively (CABI 2019a, b). In addition, more than 150 potential predator species have been listed in the literature as biocontrol agents of *B. tabaci* (Arnó et al. 2010). Nevertheless, this pest continues to cause serious damage to crops (Horowitz et al. 2011). Different types of pathogens have also been reported on these two important pest species. In order for detailed review of the research to be carried out on the biological control agents of whiteflies, they are categorized in three subsections.

4.7.2.1 Pathogens

Nearly all known pathogens of whiteflies are fungi (Table 4.1), which almost all belong to the fungal subdivision Deuteromycotina. It includes the fungi in which the "sexual phase" or "perfect stage" is unknown and hence commonly called imperfect fungi (James 2008). Although entomopathogens require specific environmental conditions that limit their success, in many countries, pathogen-based products are used to control whiteflies (Wraight et al. 2017). In case of proper and timely use of these control agents, whitefly nymphs are infected by fungi even in the absence of high-humidity conditions and will die before reaching the mature age (Vidal 2003; Fargues et al. 2005; Wraight et al. 2016). Fungi are the most frequent

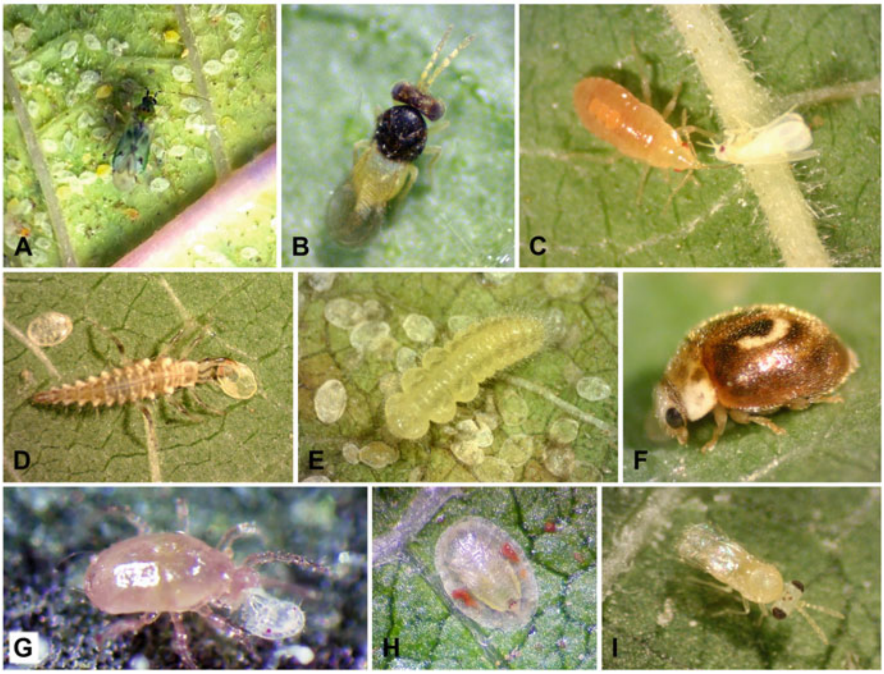

Fig. 4.5 Some predators and parasitoids of whiteflies. (**a**) *Nesidiocoris tenuis* (Reuter) (Hem.: Miridae), (**b**) *Encarsia formosa* Gahan (Hym.: Aphelinidae), (**c**) anthocorid bug (Hem.: Anthocoridae), nymph, (**d**) *Chrysoperla* sp. (Neuroptera: Chrysopidae), larva, (**e**) *Serangium* sp. (Col.: Coccinellidae), larva, (**f**) *Clitostethus arcuatus* (Rossi) (Col.: Coccinellidae), (**g**) *Amblyseius swirskii* Athias-Henriot (Acari: Phytoseiidae), (**h**) *Bemisia afer sensu lato* (Hem.: Aleyrodidae) parasitized by an *Eretmocerus* species (Hym.: Aphelinidae), (**i**) *Encarsia strenua* (Silvestri) (Hym.: Aphelinidae)

Table 4.1 Some important pathogens of the whiteflies *Bemisia tabaci* and *Trialeurodes vaporariorum*

Family	Pathogen species	Whitefly species
Clavicipitaceae	*Aschersonia aleyrodis*	*B. tabaci* and *T. vaporariorum*
Clavicipitaceae	*Aschersonia papillata*	*T. vaporariorum*
Clavicipitaceae	*Aschersonia placenta*	*T. vaporariorum*
Cordycipitaceae	*Akanthomyces lecanii*	*B. tabaci* and *T. vaporariorum*
Cordycipitaceae	*Beauveria bassiana*	*B. tabaci* and *T. vaporariorum*
Cordycipitaceae	*Cordyceps farinosa*	*B. tabaci* and *T. vaporariorum*
Cordycipitaceae	*Cordyceps fumosorosea*	*B. tabaci* and *T. vaporariorum*
Ophiocordycipitaceae	*Purpureocillium lilacinum*	*T. vaporariorum*

microorganisms (microbial) on the list of whiteflies' natural enemies (Horowitz et al. 2011). Different types of entomopathogens are the most commonly used for whiteflies in field and greenhouse crops (Perring et al. 2018). *Beauveria bassiana*

(Balsamo-Crivelli) Vuillemin, *Cordyceps fumosorosea* (Wize) Kepler, B. Shrestha & Spatafora (formerly *Isaria fumosorosea* Wize; *Paecilomyces fumosoroseus* (Wize) A.H.S. Br. & G. Sm.), *Akanthomyces muscarius* (Petch) Spatafora, Kepler & B. Shrestha (formerly *Lecanicillium muscarium* (Petch) Zare & W. Gams), *Akanthomyces lecanii* (Zimm.) Spatafora, Kepler & B. Shrestha (formerly *Verticillium lecanii* (Zimmerman) Viégas), *Metarhizium brunneum* Petch, and *Metarhizium anisopliae* (Metschnikoff) Sorokin are commercially used to control whiteflies (Gwynn 2014; Kepler et al. 2017). *Clonostachys rosea* (Link: Fr.) Schroers, Samuels, Siefert & Gams (formerly *Gliocladium roseum* Bainier) has been introduced as a control agent of *B. tabaci* (Anwar et al. 2018). The results of the recent research revealed that *C. rosea* caused 50.42% and 23.54% mortality after 6 days in fourth instar nymphs and adults of the whitefly, respectively. *Aspergillus* sp. (BC 639) has been used on transgenic cotton crops and caused 62.3% and 68.8% mortality at 14 and 21 days after treatment in nymphs and adults of *B. tabaci*, respectively (Mensah and Young 2017). Application of *C. fumosorosea* significantly reduced the population of *B. tabaci* on cucumber plants (*Cucumis sativus* L.). The first and second instars showed high susceptibility to *C. fumosorosea* with accumulative corrected mortality of 78.4% and 83.1% within 7 days, respectively (Tian et al. 2015). It has been revealed that *C. fumosorosea* CG1228 isolate caused 93% mortality on adults of *B. tabaci* biotype B, while isolates of *B. bassiana* and *C. fumosorosea* had the most effects on nymphs with 71% to 86% mortality, respectively (Mascarin et al. 2013).

The potential of using entomopathogens with some other biological agents is also a positive feature of endophytic fungi. The compatibility of *B. bassiana* with *Amblyseius swirskii* has been evaluated to control *T. vaporariorum* (Seiedy et al. 2015). There is a positive interaction between these two biocontrol agents, and they have potential to be used simultaneously (Midthassel et al. 2016). On the contrary, there is an incompatibility between *Encarsia formosa* and *B. bassiana*/*M. anisopliae* for control of *T. vaporariorum*. Of course, it is possible to use them together if entomopathogenic fungi are used a few days after parasitoid application (Hamdi et al. 2011; Oreste et al. 2016). Although some factors, such as susceptibility to fungicides, extreme dependence on environmental conditions, low shelf life, and slow performance, are all limiting factors for entomopathogens application (Faria and Wraight 2001), much research being carried out in this area in the recent years demonstrates the intention of researchers to overcome these limitations.

4.7.2.2 Parasitoids

Most of the natural enemies of the whiteflies are parasitoid wasps (Table 4.2), whose taxonomy has not yet been adequately studied, i.e., it is often extremely difficult to identify them. The whiteflies' parasitoid complex is mainly represented by seven hymenopteran families: Aphelinidae, Azotidae (formerly Azotinae, a subfamily of Aphelinidae), Eulophidae, Encyrtidae, Pteromalidae and Signiphoridae within the superfamily Chalcidoidea, and Platygasteridae in the superfamily Platygastroidea, of which the Aphelinidae provides most of the biocontrol agents (Hernández-Suárez et al. 2003; Evans 2007, 2008; Noyes 2019). Among the genera of Aphelinidae,

Table 4.2 Some important parasitoids of the whiteflies *Bemisia tabaci* and *Trialeurodes vaporariorum*

Family	Parasitoid species	Whitefly species
Aphelinidae	*Encarsia azimi*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia cibcensis*	*B. tabaci*
Aphelinidae	*Encarsia formosa*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia gennaroi*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia inaron*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia japonica*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia longifasciata*	*B. tabaci*
Aphelinidae	*Encarsia lutea*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia luteola*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia lycopersici*	*T. vaporariorum*
Aphelinidae	*Encarsia mineoi*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia nigricephala*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia porteri*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia sophia*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Encarsia strenua*	*B. tabaci*
Aphelinidae	*Encarsia tricolor*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Eretmocerus eremicus*	*B. tabaci*
Aphelinidae	*Eretmocerus haldemani*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Eretmocerus mundus*	*B. tabaci* and *T. vaporariorum*
Aphelinidae	*Eretmocerus rui*	*B. tabaci*
Platygasteridae	*Amitus arcturus*	*T. vaporariorum*
Platygasteridae	*Amitus bennetti*	*B. tabaci*
Platygasteridae	*Amitus fuscipennis*	*T. vaporariorum*

Encarsia and *Eretmocerus* include numerous species that are mostly primary parasitoids of whiteflies, and several species are of major economic importance, either as biological control agents of pest species or as components of integrated pest management programs. For example, in a review study, 112 parasitoid species have been tabulated in five families and seven genera associated with *B. tabaci* species complex, of which more than 100 species belong to *Encarsia* and *Eretmocerus* (Lahey and Stansly 2016). The parasitoid species that belong to these two genera are among the species for which almost complete physiological and ecological (population dynamics, life history, fecundity, sex ratio, etc.) studies have been carried out (Arnó et al. 2010). In the recent years, numerous articles have been published on host-parasitoid interactions of *Encarsia* and *Eretmocerus* on whiteflies (Liu 2007; Pickett et al. 2013; Schoeller and Redak 2018). Species of both genera are solitary wasps and lay their eggs inside – or in *Eretmocerus*, under – the host body. The early larval instars complete larval development within whitefly nymphs and pupate in forth instar nymphs, and then adults emerge from the latter nymphs through an exit hole cut in the dorsum of the host. Species of *Encarsia* are endoparasites; however, both sexes of *E. ectophaga* (Silvestri) on armored scales are the only case with potential of being ectoparasitoid (Hunter and Woolley 2001). Most species are autoparasitoids, with female eggs laid in a primary host and male eggs deposited as parasitoids of conspecific female or other species of *Encarsia* (Williams and Polaszek 1996; Hunter and Woolley 2001), whereas *Eretmocerus* species are ecto-endoparasitoids and both females and males develop as primary parasitoids (Horowitz et al. 2011; Lahey et al. 2016). *Encarsia formosa* Gahan prefers third and early fourth instar nymphs of *B. tabaci* and *T. vaporariorum* for oviposition, while *Eretmocerus eremicus* Rose & Zolnerowich and *Eretmocerus mundus* Mercet prefer second and third instar nymphs of *B. tabaci*. First instar nymphs are least preferred as they increase the immature period of parasitoids as well as their mortality. *Encarsia formosa* is being commercially produced and used both in inoculative and inundative release methods, in biological control programs of *T. vaporariorum* (Scopes and Biggerstaff 1971; Liu et al. 2015). It is also an effective biocontrol agent for controlling *B. tabaci*. *Eretmocerus eremicus* and *Er. mundus* have widely been used for control of *B. tabaci* (Liu et al. 2015: Perring et al. 2018), showing higher reproductive rate than *En. formosa* (Qiu et al. 2004). *Encarsia sophia*, *En. formosa*, and *Eretmocerus melanoscutus* Zolnerowich & Rose prefer later instar nymphs than younger nymphs of *B. tabaci* in greenhouses. *Encarsia sophia* feeds almost three times more than the other two species and causes 59.7% mortality in its host (Zang and Liu 2008). The efficiency of *Encarsia smithi* (Silvestri) has been evaluated on *Aleurocanthus camelliae* Kanmiya & Kasai (Hemiptera: Aleyrodidae) which is an important pest of tea plantations (Uesugi et al. 2016). The results of the recent study proved that the mortality rate of *A. camelliae* was increased using *En. smithi* and indicated its high potential as a biocontrol agent of *A. camelliae*.

4.7.2.3 Predators

The predators that have potential to control whiteflies have been thoroughly reviewed (Gerling et al. 2001; Arnó et al. 2010). Several families of insects, e.g., Coccinellidae, Anthocoridae, Miridae, Syrphidae, and Chrysopidae, as well as the mite family Phytoseiidae include the important predators (more than 150 arthropod species belonging to 9 orders and 31 families) (Table 4.3). It should definitely be noted that the list of predators and their necessary information for being used commercially are continuously changing. About 50 species of the family Coccinellidae (Coleoptera) and 22 species of Neuroptera from the families Chrysopidae and Coniopterygidae have been reported to prey on *B. tabaci* and *T. vaporariorum*. Nevertheless, most researches have been conducted on *Coccinella septempunctata* (L.), *Serangium parcesetosum* Sicard, *Nephaspis oculatus* (Blatchley), *Chrysoperla carnea* (Stephens), and *Chrysoperla rufilabris* (Burmeister), which were commercially used in integrated whitefly management programs (Deligeorgidis et al. 2005; Perring et al. 2018). The predators of whiteflies are often used in greenhouses, due to their higher efficiency on indoor crops. Most of them are generalist predators and have the ability to prey on several herbivores shifting from one to the next. In tropical greenhouses, which provide suitable environment for *B. tabaci* and *T. vaporariorum* development, this plasticity behavior

Table 4.3 Some important predators of the whiteflies *Bemisia tabaci* and *Trialeurodes vaporariorum*

Family	Predator species	Whitefly species
Aeolothripidae	*Franklinothrips vespiformis*	*B. tabaci*
Anthocoridae	*Anthocoris nemorum*	*T. vaporariorum*
Anthocoridae	*Orius niger*	*B. tabaci* and *T. vaporariorum*
Chrysopidae	*Chrysoperla carnea*	*B. tabaci* and *T. vaporariorum*
Chrysopidae	*Chrysoperla rufilabris*	*B. tabaci* and *T. vaporariorum*
Chrysopidae	*Nineta flava*	*B. tabaci*
Coccinellidae	*Clitostethus arcuatus*	*B. tabaci* and *T. vaporariorum*
Coccinellidae	*Delphastus catalinae*	*B. tabaci* and *T. vaporariorum*
Coccinellidae	*Delphastus pusillus*	*B. tabaci* and *T. vaporariorum*
Coccinellidae	*Propylea japonica*	*B. tabaci* and *T. vaporariorum*
Coccinellidae	*Serangium parcesetosum*	*B. tabaci*
Miridae	*Macrolophus caliginosus*	*B. tabaci* and *T. vaporariorum*
Miridae	*Macrolophus pygmaeus*	*B. tabaci* and *T. vaporariorum*
Miridae	*Nesidiocoris tenuis*	*B. tabaci* and *T. vaporariorum*
Phytoseiidae	*Amblyseius swirskii*	*B. tabaci*
Phytoseiidae	*Transeius montdorensis*	*B. tabaci*

of predators leads to inadequate pest control, subsequently creating inappropriate conditions in such greenhouses. Predators' particular affinity and susceptibility to cultivated plants is an important factor to use them as biocontrol agents in fields or greenhouses. For instance, although the phytoseiid mite, *Amblyseius swirskii* Athias-Henriot, is considered as one of the most effective predators and is quite active on most vegetable species, it does not have this ability on some other species, such as tomatoes (Horowitz et al. 2011). On the other hand, *A. swirskii* prefers greenhouse whitefly, *T. vaporariorum*, when this pest occurs along with the two-spotted spider mite, *Tetranychus urticae* Koch, on the strawberry plants, which shows that the diversity of pest species on host plant can increase the predator's efficiency (Mortazavi et al. 2019). The phytoseiid mites, *Amblyseius largoensis* (Muma) and *Euseius concordis* (Chant), can consume 35 and 32 eggs of the giant whitefly, *Aleurodicus cocois* (Curtis) per day, respectively (De Alfaia et al. 2018). *Aleurodicus cocois* is the main pest of the cashew trees around the world, which causes a damage of 70–90% (Goiana et al. 2017), and both abovementioned predators could be effective biocontrol agents of this pest. The predatory phytoseiid mite, *Neoseiulus cucumeris* (Oudemans), is also an effective biological control agent of *B. tabaci* and can reduce its population on greenhouse vegetables (Nawar and Erif 1993). The egg stage of whiteflies is the most susceptible stage for predation by phytoseiid mites, and mated female mites are more effective than males. The daily maximum number of eggs, first-, second-, and third-instar nymphs of *B. tabaci* killed by a single female *N. cucumeris* over a 24-h period, was calculated to be 8.5, 5.2, 3.0, and 2.1 prey compared to 6.0, 4.0, 2.3, and 1.8 prey for a single male, respectively (Li et al. 2017).

Two species of zoophytophagous mirid predators (Hemiptera: Miridae), *Macrolophus caliginosus* Wagner and *Nesidiocoris tenuis* (Reuter), are commercially produced and released for control of whiteflies, especially on greenhouse crops like tomato plants in northern Europe and Mediterranean basin (Horowitz et al. 2011). Omnivorous mirid bugs feed on both plants and pests, so they are unusual as biocontrol agents, and there has always been a lot of discussion about the use of these species. Nevertheless, *Macrolophus pygmaeus* (Rambur) is used for more than 20 years and is considered harmless to many crops including tomatoes. Although it feeds on reproductive organs of tomato plants and provides "disservice" to the crop, tomato plants with a lower number of *M. pygmaeus* have shown to have a significantly higher number of whiteflies than those with a higher number of the predator, and vice versa (Sanchez et al. 2018). Mean fertility of *M. pygmaeus* females is usually higher when feeding on *B. tabaci* nymphs than when feeding on the potato aphid, *Macrosiphum euphorbiae* (Thomas), and eggs of the Mediterranean flour moth, *Ephestia kuehniella* Zeller, and the tomato leafminer, *Tuta absoluta* (Meyrick), which shows higher tendency of the predator to feed on *B. tabaci* (Sylla et al. 2016). Further research on another omnivorous mirid predator, *N. tenuis*, is also still ongoing. Given that the calculated degree day and estimated upper temperature threshold for egg-adult of the predator was less than sweet potato whitefly (*B. tabaci*) (340 vs. 390 DD; ≈35 °C vs. ≈ 40 °C, respectively), *N. tenuis* could be used as a biological agent against the whitefly on solanaceous crops in early season (Mirhosseini et al. 2018). Larvae of the western flower thrips, *Frankliniella occidentalis* (Pergande) (Thysanoptera: Thripidae), are another omnivore that prey on *T. vaporariorum* crawlers on cucumber (*Cucumis sativa* L.) and sweet pepper (*Capsicum annuum* L.) (high- and poor-quality host plants, respectively) although they do not feed on the whitefly eggs. In the presence of greenhouse whitefly, the development and oviposition rate of *F. occidentalis* have been higher than when the thrips fed on cucumber leaves without whitefly crawlers (Van Maanen et al. 2012).

4.7.2.4 Application of Multiple Natural Enemies

The use of multiple natural enemies raises a question of how the interactions between the different biological control agents that are competing for the same pray affect their efficacy. As defined by Ferguson and Stiling (1996), when more than one natural enemy is used, they may act synergistically, additively, or antagonistically. Therefore, the compatibility of multiple natural enemies and the occurrence of intraguild interactions between them need to be studied prior to their combined application in biological control programs (Fathipour and Maleknia 2016).

A combination of natural enemies can influence observed rate of predation and parasitism, and depending on what the objectives are, it can be used for particular biological control programs. For instance, a mixed release of one of the two whitefly parasitoids, namely, *E. formosa* and *E. sophia*, along with the polyphagous predatory ladybird *Harmonia axyridis* (Pallas) (Coleoptera: Coccinellidae) on sweet potato whitefly minimizes the predation rate of *H. axyridis*. Using the predator with both parasitoids together improves the predation rate of the predator

(Tan et al. 2016). Interestingly, the predator prefers non-parasitized nymphs of the whitefly as prey. The number of parasitized whiteflies increases when all the three biocontrol agents are introduced rather than when the wasps are released either alone or mixed with the other parasitoid. The adult parasitoid emergence is significantly reduced in the presence of *H. axyridis* in comparison with releasing each parasitoid alone. Therefore, the best option to establish these parasitoids would be only to release them individually or together; however, introducing one of the parasitoids along with the predator would be the best choice, if the whitefly reduction is the only aim of the control program. The latter causes the increase of parasitism rate and preference of the predator for non-parasitized nymphs. Such information might be important in enhancing the effectiveness of biological control programs (Tan et al. 2016).

Macrolophus melanotoma (Costa) (Hemiptera: Miridae) is not successful against *B. tabaci* on greenhouse eggplants when released alone, but in combination with the parasitoid *Er. mundus*, it will improve the success of the whitefly control program (Karut et al. 2018). Parasitism capacity of *E. formosa* is usually reduced during the winter in greenhouses (Gerling et al. 2001), for which the climate parameters, high infestation level, and severe honeydew production by whiteflies can be acceptable reasons. The two predatory mites, *Amblydromalus limonicus* (Garman & McGregor) and *Transeius montdorensis* (Schicha) (Acari: Phytoseiidae), can be a good supplement or even alternative to *E. formosa* for control of *B. tabaci* and *T. vaporariorum* (Richter 2017) because of their ability to overcome poor weather conditions and use alternative food sources.

4.7.3 Chemical Control

4.7.3.1 Synthetic Pesticides and Biopesticides

Over the years, chemical insecticides have been the key tool to control whiteflies in greenhouses and fields, and that is why there is a lot of research about the positive and negative features and effects of this control measure. Using synthetic insecticides has been the usual method for control of whiteflies in many parts of the world. Since most synthetic pesticides have broad-spectrum activity against nontarget organisms including natural enemies, their effects should be tested on local dominant species of natural enemies. In most cases, not only lethal doses but also sublethal doses of synthetic pesticides may adversely affect population parameters of whiteflies as well as those of its natural enemies, and this trade-off should be taken into consideration in any pesticide application program. During about a century, various classes of insecticides including Pyrethroids, Organophosphates, Carbamates, and Organochlorines were used to fight against whiteflies (Horowitz et al. 2011). Hydrocyanic acid gas, DDT, and parathion were the earliest of these insecticides, and bifenthrin, buprofezin, imidacloprid, fenpropathrin, amitraz, fenoxycarb, deltamethrin, azadirachtin, and pymetrozine are the compounds that are being used later and to this day (Smith 1970; Dennehy et al. 2010). However, it is noteworthy that some of the insecticides mentioned above are used to suppress virus transmission by whiteflies, especially *B. tabaci*; of course, they have not been successful in many cases (Henneberry 1993). Insects' resistance to insecticides is increasing, and their use cannot be a promise solution to accurately control whiteflies on agricultural crops and ornamental plants. Whiteflies have highly polyphagous nature, and the easy dispersion of adults by wind and underleaf habitat of immature stages and adults are some of the reasons for the resistance to insecticides (Horowitz et al. 2011). However, new insecticides including nicotinic receptor agonists, insect growth regulators (IGRs), cyantraniliprole, ryanodine receptor insecticides, spiromesifen, and spirotetramat can be used as one of the components of an integrated pest management (IPM) program due to their lower environmental risk and other specific and positive characteristics (Perring et al. 2018). The efficacy of some insecticides on *T. vaporariorum* in field and greenhouse-grown strawberry plants has been determined (Bi et al. 2002). The results of the recent research reveal that imidacloprid, pyriproxyfen, buprofezin, and pyridaben could be suitable controllers for whiteflies on strawberries, especially in greenhouses. The soil-applied imidacloprid caused 82–96% mortality on the whitefly in the greenhouse. An application of buprofezin followed by pyriproxyfen decreased nymphal density of the whitefly by 61–85% in strawberry fields. The sublethal and low-lethal concentrations of imidacloprid and bifenthrin caused phloem feeding (most important feeding trait in a plant protection perspective) and honeydew excretion reduction and subsequently fecundity decrease of the tobacco whitefly (*B. tabaci*). Indeed, they have an antifeedant effect, which indirectly does not allow *B. tabaci* to develop and reproduce (He et al. 2013). Changes in insect behavior and physiology are the effects of the sublethal concentrations of insecticides (Desneux et al. 2007). It has been revealed that sublethal concentrations of Calypso (neonicotinoid insecticide) caused reduction in longevity and fecundity of females, net reproductive rate (R_0), intrinsic rate of increase (r), and significantly increased mean generation time (T) in the greenhouse whitefly (Safavi and Bakhshaei 2017). The use of this insecticide is appropriate in cucumber and tomato greenhouses according to its characteristics in reducing the chemical residuals. The sublethal [LC_{10} (0.22 mg/L) and LC_{25} (0.63 mg/L)] concentrations of cyantraniliprole on *B. tabaci* MED (Mediterranean) which is threatening agricultural production in China because of its resistance to the different insecticides reduced fecundity of females, hatching rate of eggs laid by females, oviposition duration, and survival rate of nymphal stages and also increased nymphal developmental time and decreased adult longevity of the whitefly (Wang et al. 2017). The authors also examined the effect of seven other compounds that include clothianidin, dinotefuran, acetamiprid, nitenpyram, imidacloprid, thiamethoxam, and chlorantraniliprole, and their toxicity was 1.8, 1.9, 2.7, 3.0, 5.9, 6.9, and 349.0 times less than cyantraniliprole, respectively. The mentioned features indicate that cyantraniliprole is an appropriate alternative to control *B. tabaci* MED (Wang et al. 2017). It has been proved that *B. tabaci* adults do not feed on the plants treated with cyantraniliprole, which help agricultural crops to be protected from whitefly attacks and diseases transferred by it (Civolani et al. 2014). Using the combination of two insecticides can also be a solution to increase the effectiveness of chemical control. Application of thiacloprid with deltamethrin decreased the fertility percentage and the survival rate of nymphs and adults of sugarcane whitefly, *Neomaskellia andropogonis* Corbett. Although this combination can have a beneficial effect on reduction of the pest population, its harmful effect on parasitism rate of *Encarsia inaron* and *Eretmocerus delhiensis* is not unexpected (Behnam-Oskuyee et al. 2018).

Biopesticides, also known as biological pesticides, are derived from natural materials such as animals, plants, and bacteria. The advantages of most biopesticides over synthetic compounds include their lower toxicity to nontarget organisms and their quick decomposition. Most biopesticides target specific pests and generally pose little or no risk to humans or the environment. Plant-based products and their chemical components are suitable and low-risk alternatives for using in IPM programs, because of their repellency, contact and fumigant toxicity, regulation of growth, and pathogenic properties (Murray 2000). Oils also kill insects by suffocation, while they are environmentally nonpersistent and safe for humans (Stansly et al. 1996; Wagan et al. 2017). Various insecticides were evaluated in a 2-year (2011 to 2013) research project in tomato fields of India for *B. tabaci* control. The combination of neem and *Spilanthes* caused 62.39% suppression, while individual application of them resulted in 51.72% and 54.92% suppression, respectively. This positive change by all three of them was also observed in the 2-year yield of the product (27.75, 25.56, and 26.67 tons/hectare, respectively). However, in the mentioned study, acetamiprid had the highest efficiency with 76.59% suppression, and tobacco and garlic extract had the lowest level of suppression by 43.74% and 37.36%, respectively. The yield of the product when using acetamiprid was 30.15 t/ha, while this amount was reduced to 24.02 and 23.11 t/ha for tobacco and garlic extract, respectively. Although the plant extracts had a lesser impact on the population of insect pest due to the high toxicity of acetamiprid, it makes us convinced that using plant extracts because of benefits we receive regarding higher human health lowers environmental hazard and achieves organic farming (Subba et al. 2017). In a recent research in China, the repellency and anti-oviposition effects of the essential oils of four plant species (*Curcuma longa*, *Litsea cubeba*, *Piper nigrum*, and *Zanthoxylum bungeanum*) have been investigated on *B. tabaci* for the first time. All of them have strong repellency against whitefly and deter oviposition, especially essential oils from *L. cubeba*. This compound has 69.14% and 54.77% repellency for adult females at 24 h in laboratory and greenhouse, respectively (Wagan et al. 2017). *Gardenia jasminoides* J. Ellis and its four main chemicals can be considered as effective plant extracts on whiteflies' control by affecting oviposition capacity. The essential oil treatment in fumigation causes 81.48% and in contact causes 46.44% toxicity against whitefly adults and nymphs, respectively. These compounds have other positive effects in pest control programs. They also show 89.59% and 80.08% repellency and 63.58% anti-oviposition activity against whitefly adults and nymphs at 24 h. LC_{50} of them for whitefly is at least 2396.46 ppm (Wagan et al. 2018). It has been demonstrated that the aqueous extracts of *Ruta chalepensis*, *Peganum harmala*, and *Alkanna strigosa* have substantial insecticidal activity against *B. tabaci* immature stages, and they are suitable components for IPM programs, although *A. strigosa* negatively affects parasitoids' population (Al-Mazra and Ateyyat 2009). The essential oil of *Laurelia sempervirens* (Ruiz & Pav.) Tul. leaves is also effective on *T. vaporariorum*, although they are also toxic to its parasitoids (Zapata et al. 2016). Therefore, the side effects of essential oils on predators and parasitoids should be taken into consideration in their use in whitefly IPM programs.

4.7.3.2 Whitefly Resistance to Pesticides

Insecticide resistance is the most important barrier against insecticide-based control programs. This is a universal problem for all pests. Whiteflies, depending on environmental conditions and host plant factors, may also exhibit ecological (polyphagy, dispersal, r-selection, adaptability), physiological, and morphological characteristics that enable them to develop resistance to synthetic chemical compounds. Progress in preventing resistance to whiteflies requires appropriate information on these characteristics. Frequent insecticide application, which has always been the easiest solution to control these noxious pests, in addition to resistance problems (Castle et al. 2010), causes the loss of balance in the resistant and susceptible populations of the pest as well as the destruction of natural enemies (Eveleens 1983). Different genes and enzymes help whiteflies to develop resistance mechanisms with enhancing metabolic detoxification and target-site insensitivity. The cytochrome P450 monooxygenase (P450), s-transferase (GST), and carboxylesterase (COE) can be the reason of resistance to diafenthiuron in *B. tabaci*, biotype B, due to being significantly higher in resistant strains compared with susceptible ones (Zhang et al. 2016). The gene expression of these enzymes is also significantly higher in the resistant strain than the susceptible one. Resistance to organophosphates has also been reported in whiteflies caused by these enzymes (Rauch and Nauen 2003; Alon et al. 2008; Yang et al. 2013). In the case of observing resistance responses on a molecular scale, it might be helpful to use ecological scales to understand how to deal with this issue.

Identification of resistance mechanisms is important to prevent and manage whitefly resistance to insecticides and requires further research. Four resistance mechanisms have been proposed as follows (Fathipour and Mirhosseini 2017):

4.7.3.2.1 Metabolic Resistance

In this mechanism, resistant insects may detoxify or destroy the toxin faster than susceptible insects or prevent the toxin from reaching target sites by binding it to proteins or other compounds in their bodies. This kind of resistance is the most common mechanism and often presents the greatest challenge to its avoidance. This detoxification method involves three phases: recognition and hydrolysis of the toxin, conversion of toxin into a nontoxic molecule, and excretion of the molecule from the insect's body.

4.7.3.2.2 Altered Target-Site Resistance

The target site where insecticides usually bind in insects can become modified to reduce insecticide effects. This is the most common mechanism of resistance in fungi and weeds, and it is also prevalent in insects.

4.7.3.2.3 Behavioral Resistance

Resistant insects may avoid the negative effects of insecticides by changing their normal activity patterns. Insects may stop feeding or move to the underside of the sprayed leaf. Resistance can be accelerated by the deliberate mating of resistant males and females. Using different insecticides with different modes of action can help reduce the selection pressure for resistance.

4.7.3.2.4 Penetration Resistance

In this case, resistant insects may absorb the toxin more slowly than susceptible individuals. Penetration resistance occurs when the outer cuticle develops barriers that can slow absorption of insecticides. This kind of resistance is frequently present in combination with other types of resistance and can protect insects from a wide range of insecticides.

4.7.3.3 Whitefly Resistance Management Strategies

4.7.3.3.1 Minimizing Insecticide Use

Minimizing insecticide use is fundamental to pesticide resistance management. From an insecticide resistance viewpoint, every time an insect population is treated with an insecticide, selection for resistance is occurring. Although this selection is not evident at first because so few individuals survive, as the resistant population increases, nymphal survival and subsequent plant damage becomes obvious. Intensification of spray programs can exacerbate the problem by putting even more resistance selection pressure on whitefly populations. Determination of the economic injury level is important, because trying to produce a crop with a zero tolerance for damage may work in the short term but leads to increased insecticide use, insecticide resistance and, ultimately, loss of pest control, and increased crop loss. The use of nonchemical strategies such as crop rotation (host-free periods), biological control, and weed control can help reduce the need for chemicals and consequently slow the development of insecticide resistance (Fathipour and Mirhosseini 2017).

4.7.3.3.2 Mixtures of Insecticides with Different Modes of Action

Mixing insecticides with different modes of action or different mechanisms of resistance can be effective in managing resistance development. Two insecticides and a suitable synergist is a typical combination. Insecticides with similar modes of action generally express cross-resistance (selection for resistance to one product selects for resistance to all products with the same mode of action). However, long-term use of mixtures of different classes of insecticide can also give rise to insecticide resistance, if resistance mechanisms to different insecticides arise together in some individuals. Continued use of the mixture will select for these multiple-insecticide-resistant pests.

4.7.3.3.3 Using Insecticides with Short Persistence

Whiteflies with resistant genes will be selected over susceptible ones whenever insecticide concentrations kill only the susceptible individuals. An ideal insecticide quickly disappears from the environment so that persistence of a “selecting dose” does not occur. In contrast, environmental pollution, damage to natural enemies, and increasing risk for consumers are all intensified by using persistent insecticides.

4.7.3.3.4 Rotations of Insecticide

The rotation or alternation of insecticides is another tactic used to manage resistance development. Longer use of a single insecticide class will enhance the chance of resistance (especially in multivoltine insects such as whiteflies) since the survivors of the first generation and those of the next generations will most likely be tolerant to that class. In this tactic, it is important that pest resistance to the two alternatively being used insecticides be rare, that the alternating insecticides belong to unrelated chemical classes, and that the time interval between applications of the rotating insecticides is long enough for the pest population to return to its original level of susceptibility.

4.7.4 Host Plant Resistance

Host plant resistance strategy, which may also be integrated with other control methods, can play an important role in management programs as low-cost, long-term, practical solution and the last line of plant defense against herbivores. This method can also act as an alternative management strategy for whiteflies by making changes in their biology, physiology, or even their behavior. Although plant and herbivore interactions are very complex and multifaceted, comprehensive information on genetic resistance mechanisms in plants offers the power to choose a resistant variety among different varieties of plants to minimize the impact of the pest (Rasmann and Agrawal 2009; Taggar and Gill 2016).

Host plant resistance can be an effective replacement for broad-spectrum insecticides. Potentially, deployment of resistant crop varieties is inexpensive, easy to introduce, specific to one or several pests, persistent, easy to integrate with other farm operations, compatible with other IPM control tactics, and, more importantly, environmentally safe. While such resistance is based on heritable traits, some of these traits may fluctuate widely under different environmental conditions. Accordingly, host plant resistance may be classified as either genetic (traits which are under the primary control of genetic factors) which is very popular in IPM programs or as ecological (traits which are under the primary control of environmental factors). Genetic resistance includes both induced resistance (biotic and abiotic environmental factors which lower insect fitness or negatively affect the pest’s host selection processes) and constitutive resistance (inherited characteristics whose expression, although influenced by the environment, is not triggered by environmental factors) (Metcalf and Luckmann 1994). Mechanisms of genetic resistance include antixenosis, antibiosis, and tolerance (Fig. 4.6), which are evaluated using different methods (Smith et al. 1994). Host plant resistance to pests can be inherited either vertically (controlled by a single gene) or horizontally (controlled by many genes). It is much more difficult to breed cultivars with horizontal resistance to insect pests

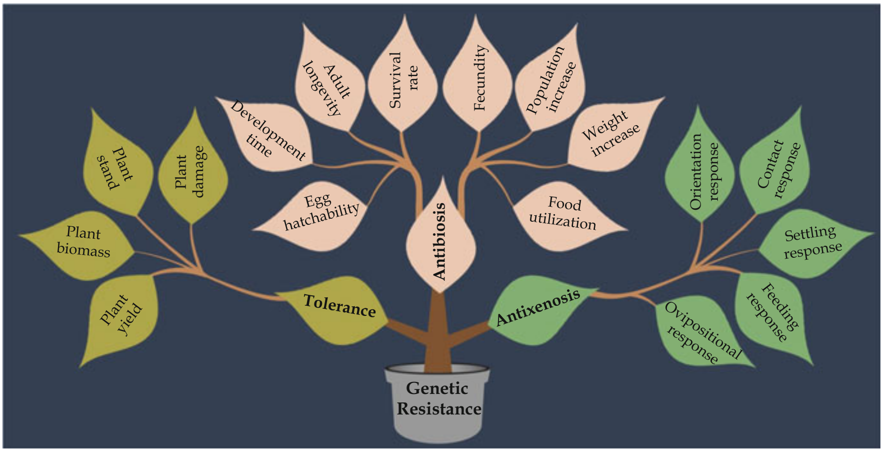

Fig. 4.6 Genetic resistance mechanisms identified in host plants and their evaluation methods

because of the large number of genes involved. To determine which mechanism is active in particular cases, experiments can be designed that assess the pest's life table parameters among varieties (Chi and Liu 1985; Chi 1988; Fathipour and Maleknia 2016) (Table 4.4), as well as their nutritional indices (Waldbauer 1968; Fathipour and Mirhosseini 2017) (Table 4.5), and the pest's digestive proteolytic and amylolytic activities (Fathipour and Naseri 2011).

The four stages, including screening, categorization, breeding, and implementation, have traditionally been used for host plant resistance application in IPM programs (Stout and Resistance 2014). In a large-scale screening, 550 cotton genotypes resistant to *B. tabaci* have been evaluated by Zhu et al. (2018) in their experiments. They observed that the leaves with higher density of hairs (trichomes) had less number of eggs/nymphs of the whitefly, which indicated their importance for antixenotic resistance. Two genotypes of common bean (*Phaseolus vulgaris* L.) Arc 1 and IAC Alvorada have shown a relatively high resistance to *B. tabaci*. They increased mortality of whitefly nymphs twofold more than other genotypes, by 44–78% and 46–96.7% mortality, respectively (Hoshino 2017). The researchers attributed this result to mechanical and/or antibiosis resistance. Wild cultivar of tomato plant *Lycopersicon pimpinellifolium* is also more resistant than commercial cultivars (*Solanum lycopersicum* var. 'Elegance') to oviposition and feeding of *T. vaporariorum* (Mcdaniel et al. 2016). The characteristics of trichome have a key role in host plant selection of *T. vaporariorum* for oviposition (Maliepaard et al. 1995; Avery et al. 2015).

Table 4.4 Equations and concepts of female-based and two-sex-based life table parameters

Female-based life table	Two-sex-based life table
x (day) Age	x (day) Age
N_x Number of surviving individuals (only females in adult stage) entering the age x	N_x Number of surviving individuals (females and males in adult stage) entering the age x
M_x Daily mean number of eggs produced per female of age x	f_{xj} Age-stage-specific fecundity (daily number of eggs produced per female of age x)
$l_x = \frac{N_x}{N_0}$ The age-specific survival; N_0 = number of individuals at the age $x = 0$	s_{xj}; $l_x = \sum_{j=1}^{k} s_{xj}$ Age-stage-specific survival rate (x = age and j = stage); k = number of stages
m_x Daily mean number of female eggs produced per female of age x	$m_x = \frac{\sum_{j=1}^{k} s_{xj} f_{xj}}{\sum_{j=1}^{k} s_{xj}}$ Age-specific fecundity (daily number of eggs produced per individual, i.e., this number is divided by all individuals [males and females] of age x); k = number of stages
$R_0 = \sum_{x=\alpha}^{\beta} l_x m_x$ The net reproductive rate (female eggs per female)	$R_0 = \sum_{x=0}^{\omega} \sum_{j=1}^{k} s_{xj} f_{xj}$ The net reproductive rate (eggs per individual)
$\sum_{x=0}^{\omega} e^{-r(x+1)} l_x m_x = 1$ Intrinsic rate of increase (r) (number of females added to the population per female per day, i.e., the intrinsic birth rate (b) minus the intrinsic death rate (d)) (day^{-1})	$\sum_{x=0}^{\omega} e^{-r(x+1)} l_x m_x = 1$ Intrinsic rate of increase (r) (number of individuals added to the population per individual per day, i.e., the intrinsic birth rate (b) minus the intrinsic death rate (d)) (day^{-1})
$\lambda = e^r$ Finite rate of increase (the rate at which the population [only females] increases from 1 day to the next day) (day^{-1})	$\lambda = e^r$ Finite rate of increase (the rate at which the population [females and males] increases from 1 day to the next day) (day^{-1})
$GRR = \sum_{x=\alpha}^{\beta} m_x$ The gross reproductive rate (female eggs per female)	$GRR = \sum_{x=\alpha}^{\beta} m_x$ The gross reproductive rate (eggs per individual)
$T = \frac{\ln R_0}{r}$ Mean generation time (day)	$T = \frac{\ln R_0}{r}$ Mean generation time (day)

4.7.5 Integrated Management of Whiteflies in Indoor and Outdoor Cropping Systems

As described in detail in this chapter, whiteflies are destructive pests in both fields and greenhouses worldwide. For many years, farmers have been spending a lot of money to control different species of whiteflies in both indoor and outdoor cropping

Table 4.5 Nutritional indices for determining consumption and utilization of food by insects

Nutritional index	Equation
Relative consumption rate (RCR) or consumption index (CI) (unit: Mg/mg/day)	$\frac{\textit{Fresh or dry weight of food eaten (ingested) during feeding period (mg)}(E)}{\textit{Mean fresh or dry weight of insect during feeding period (mg)}\ (A)\times\textit{duration of feeding period (day)}(T)}$ $\frac{E}{A\times T}$
Relative growth rate (RGR) (unit: Mg/mg/day)	$\frac{\textit{Fresh or dry weight gain (increase) of insect during feeding period (mg)}(G)}{\textit{Mean fresh or dry weight of insect during feeding period (mg)}(A)\times\textit{duration of feeding period (day)}(T)}$ $\frac{G}{A\times T}$
Efficiency of conversion of ingested food (ECI) (unit: percent)	$\left[\frac{\textit{Fresh or dry weight gain (increase) of insect during feeding period (mg)}(G)}{\textit{Fresh or dry weight of food eaten (ingested) during feeding period (mg)}(E)}\right]\times 100$ $\left(\frac{G}{E}\right)\times 100$ or $\left(\frac{RGR}{RCR}\right)\times 100$
Efficiency of conversion of digested food (ECD) (unit: percent)	$\left[\frac{\textit{Fresh or dry weight gain (increase) of insect during feeding period (mg)}(G)}{\textit{Fresh or dry weight of food eaten (ingested) during feeding period (mg)}(E)- \textit{Fresh or dry weight of feces produced during feeding priod (mg)}(F)}\right]\times 100$ $\left(\frac{G}{E-F}\right)\times 100$
Approximate digestibility (AD) (unit: percent)	$\left[\frac{\textit{Fresh or dry weight of food eaten (ingested) during feeding period (mg)}(E)- \textit{Fresh or dry weight of feces produced during feeding period (mg)}(F)}{\textit{Fresh or dry weight of food eaten (ingested) during feeding period (mg)}(E)}\right]\times 100$ $\left(\frac{E-F}{E}\right)\times 100$

systems. Similarly, entomologists have also introduced some effective methods for control of whiteflies (Fig. 4.7). They also determined the advantages and disadvantages of combining different control measures and offered important prescriptions for management of these noxious pests, which certainly were diverse for different crops and environments. Despite all of the abovementioned items, farmers still have little interest in setting up management plans for whiteflies. This resulted in high application of chemical insecticides against whiteflies, especially *B. tabaci* and *T. vaporariorum*, and consequently increasing resistance of these two pests to a wide range of insecticides. It should not be overlooked that most management plans are limited to *B. tabaci* species complex and *T. vaporariorum* and other species and their different cultivating environment have been neglected. In this section, we have tried to discuss different strategies and methods involved in the integrated management of greenhouse and field whiteflies separately. As a general and practical point of view regarding chemical control in both environments, the following items should be considered: (i) use of bioinsecticides instead of synthetic ones, (ii) use of selective insecticides instead of broad-spectrum ones, (iii) minimizing the use of insecticides, (iv) mixing insecticides with different modes of action, (v) using insecticides with short persistence, and (vi) rotation of insecticides (Fathipour and Mirhosseini 2017).

Trialeurodes vaporariorum and *B. tabaci* are the most harmful whiteflies in greenhouses. Although there are other active whiteflies in this environment, the focus usually is on these species because of their higher importance in terms of economic damage. *Bemisia tabaci* damages tomatoes, beans, cucurbits, and pepper products in greenhouses and transmits viruses, so the combination of different control methods is very important for its suppressing. *Trialeurodes vaporariorum* feeds on approximately 859 plant species such as eggplant, cucumber, beans, sweet peppers, and tomatoes belonging to 469 genera in 121 families (CABI 2019b). Seedling stage and vegetative growing stage of crops are attacked by *T. vaporariorum*. The honeydew and sooty mold in fruits and stems, abnormal color, abnormal form and necrotic area in leaves, and dwarfing of plants are the most reminiscent symptoms of the greenhouse whitefly. The appropriate monitoring is the first step for any pest management programs, and pan counts, vacuum samples, visual sampling (leaf turning), and yellow sticky traps are different ways of monitoring this pest. The monitoring should be started from the very early stages of plant growth. Of course, these conditions vary from plant to plant, for example, in a tomato plant, the most appropriate start time for monitoring is when the plant has about five leaves (Stansly et al. 2010). For a closer inspection at the greenhouse environment and identification of the pest species (detection between greenhouse whitefly and tobacco whitefly), a x10 hand lens can be used to look for suspicious plants that seem stunted or chlorotic on both surfaces of leaves. This activity should be repeated weekly throughout the growing season in order to identify the growing rate of whitefly population. In the control measures section, there are many examples for each whitefly control method in different environments such as greenhouses. The combination of cultural, biological, and chemical control measures can work in line with IPM and consider the health of the product, human, and environment.

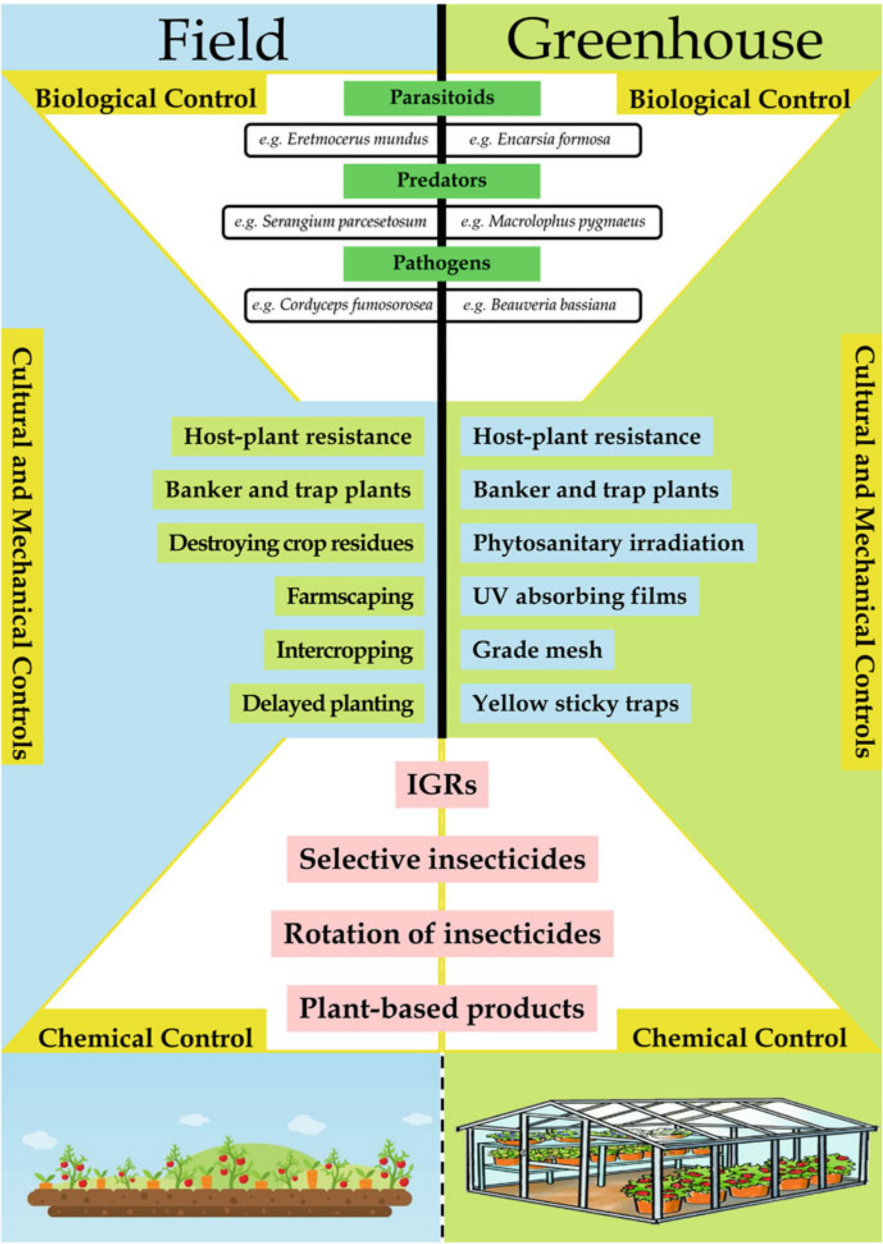

Fig. 4.7 Some effective methods for control of whiteflies in indoor (greenhouses) and outdoor (fields) cropping systems

Therefore, we have tried to make successful examples of the combination of methods in the following text. The regular removal of weeds around the greenhouse doors, which is the main location of whitefly population, would be an efficient tactic for reduction of initial population density of whiteflies. The installation of physical obstacles, such as grade mesh with pore size of 400 microns that prevents entrance of whiteflies to the greenhouse, and cleaning the production area and removing all plants and weeds that can be contaminated with whiteflies after crop harvest are the most common cultural control methods in greenhouses. The presence of UV-absorbing films and appropriate netting in the greenhouse itself is also beneficial for pest control, especially for whiteflies. The parasitoid wasps of *E. formosa* and *Er. eremicus,* the green lacewing of *Mallada signata* (Schneider), the phytoseiid predatory mites of *Typhlodromips montdorensis* (Schicha) and *Amblyseius swirskii,* and some of entomopathogens are number of the most effective biocontrol agents that might be used in whitefly management programs. Regarding the insecticides, it should be considered that the use of selective insecticides, alternate classes of insecticides, and soaps and oil sprays prevents resistance developing. Various types of insecticides are used against whiteflies in greenhouses. The most widely used ones are imidacloprid (Zhu and Ju 1990), buprofezin, and pymetrozine, which are one of the arms of management programs. In some management plans to control the greenhouse whitefly, the following compounds are used: azadirachtin, insecticidal soap, neem oil, and carbaryl. The mentioned arrangement plan is based on the most effective and least harmful insecticides to natural enemies, honey bees, and the environment. The use of imidacloprid along with *E. inaron* in management plans reduces population density and damage of *T. vaporariorum*. This combination causes 97.2% mortality in population of immature whiteflies. Entomopathogens and plant oils are the other components, in which their combination has positive performance in whitefly control strategies. The individual application of Tri-Tek (a petroleum horticultural oil) and *B. bassiana* provided 100% and 74% egg mortality and 69% and 65% second instar larvae mortality on Mediterranean species of *B. tabaci* species complex, respectively. But the tank-mix application of them caused 95.5% mortality of second instar larvae under greenhouse conditions, which represents their high efficiency in controlling this dangerous and important pest (Cuthbertson et al. 2015). These examples of biological (parasitoids or entomopathogens) and chemical control integration demonstrate their better performance when used simultaneously compared with individual use against the greenhouse whitefly, as well as their ability to be the components of IPM programs.

The tobacco whitefly (*B. tabaci*) is the most important whitefly on field crops including cassava, cotton, sweet potatoes, tobacco, and tomatoes. Although some of its biotypes feed on a few plants, the MEAM1 biotype is obviously polyphagous. The pupae and adults of *B. tabaci* feed on the flowering, seedling, and vegetative growing stages of plants. Many components, which can be used to run an IPM program on whitefly-infested fields such as biological control, host plant resistance, selective insecticides, and cultural, mechanical, and physical methods, have previously been explained in this chapter. Therefore, in this section, we will provide successful examples of their combination in a framework of IPM programs. The key points that should be taken into consideration in management plans of this pest are as follows:

1. Chemical insecticides are harmful to natural enemies, human health, and environment. Furthermore, reports of pest resistance are increasing. Can they be dropped? Absolutely not. Therefore, these compounds should be considered as an effective component of IPM; however, newer and less harmful insecticides should be used.
2. If biocontrol agents used alone cannot be effective in whitefly control programs, tendency of farmers to use these agents will be much lower than the chemical insecticides. Therefore, in most cases, a combination of these two major control measures would be more effective and popular. Since the tobacco whitefly is economically important in terms of causing damage on valuable crops or transmitting the plant viruses, it needs to be controlled by appropriate combination of different control measures even those with less importance like the mechanical and physical methods or those with less effectiveness like host plant resistance. Finally, using all of these methods in the management plans with area-wide or community-based management approach, in addition to having specific benefits in crop production, it can also reduce the environmental concerns.

One of the most interesting experiences with the integration of control methods in management programs of whitefly pests is the combination of entomopathogenic fungi with the plant-based or chemical insecticides. Separate combination of buprofezin, imidacloprid, and nicotine (plant extracts) with *Akanthomyces muscarium* as entomopathogenic fungi has shown high mortality on *B. tabaci* (68.5, 89.2%, and 68.9, respectively) (Cuthbertson et al. 2005). The high level of control occurs when the insecticides are applied at first; then the fungus is used. The entomopathogenic fungus *Isaria javanica* in combination with the sublethal concentration (LC_{25}) of spiromesifen and buprofezin can be used in IPM systems of whiteflies. They have in vitro compatibility, and the germination and mycelial growth of *I. javanica* is not affected when insecticides are used. The application of fungus alone on whiteflies causes up to 62.4% nymphal mortality, while its combinations with buprofezin or spiromesifen increase its efficiency by 10% and 24%, respectively. In addition to the use of chemical compounds, many researchers have proposed the combination of plant derivatives and fungi. There is a strong synergistic relationship between matrine (a plant-based insecticide, derived from the roots of *Sophora flavescens* and *S. alopecuroides* (Fabaceae)) and an entomopathogenic fungus *A. muscarium*. Their combination negatively affects enzyme activity of *B. tabaci* such as acetylcholinesterase, carboxylesterase, catalase, chitinase, glutathione-s-transferase, peroxidase, and superoxide dismutase and finally increases the mortality rate of the whitefly. This effect could probably be due to the disturbance of acetylcholine balance as target receptors of individual component of the combination (Ali et al. 2017). Combined use of *B. bassiana* (NATURALIS) with *Calotropis procera* (Apocynaceae) or *Inula viscosa* (Asteraceae) extracts and also combined use of *Metarhizium brunneum*

(BIPESCO5) with *C. procera* or *I. viscose* extracts cause 65.2%, 56.2%, 41.7%, and 44.3% mortality in *B. tabaci* adults, respectively, while individual application of *B. bassiana* and *M. brunneum* only kills 26.1% and 18% of adults, respectively. These results show the importance of using different control factors together. Accordingly, endophytic entomopathogenic fungi and aqueous plant extracts are compatible and may effectively contribute to an integrated whitefly management program when applied together. However, virulent endophytic fungal strains should be combined with the most effective plant extract to achieve higher efficacy levels (Jaber et al. 2018).

In field management plans of whitefly pests, the impact of chemical insecticides on predators and parasitoids, as well as the compatibility of these two main control measures, should be taken into consideration. The use of biocontrol agents, especially parasitoids, is a key point in integrated pest management programs, but it seems that their use alone cannot dramatically reduce whitefly population. At present, chemical insecticides are the most widely used control method in most parts of the world, which decreases the real activity and potential of natural enemies, especially parasitoids. Using the safe and selective pesticides is an appropriate solution for letting the natural enemies show their real role in natural control of the noxious pests, like whiteflies.

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Mealybugs

5

S. Subramanian, T. Boopathi, Suresh M. Nebapure, Yogesh Yele,
and K. Shankarganesh

Contents

Abstract

The mealybugs are soft-bodied sap-sucking insect pests catalogued under the group of scale insects (Hemiptera: Coccoidea) with the worldwide distribution of about 2000 species. Often, mealybugs are mistaken as woolly aphids, cotton cushion scale and even some whiteflies and small scales owing to their waxy coat, producing honeydew accompanied with sooty mould. They are called as

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S. Subramanian (✉) · S. M. Nebapure
ICAR – Indian Agricultural Research Institute, New Delhi, India

T. Boopathi
ICAR – National Bureau of Plant Genetic Resources, New Delhi, India

Y. Yele
ICAR – National Institute of Biotic Stress Management, Raipur, India

K. Shankarganesh
ICAR – CICR, Regional Station, Coimbatore, India

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mealybugs because of the mealy wax secretion all around their bodies. Mealybugs are serious insect pests of diverse cropping systems the world over with over 160 species of mealybugs being labelled as crop pests. These pests are often considered as global invasive pests on account of their wider distribution and quick spread and due to their polyphagous status. Mealybugs are phloem feeders, and the characteristic leaf-curling damage by them might be the result of calcium extraction linked to pectin degradation by salivary pectin esterases. Many of the mealybug pests also act as vectors for various virus diseases. They are hard to be controlled by insecticides due to its morphological, structural and behavioural adaptability with the tendency to aggregate and settle themselves in cracks or crevices in tree barks, under the leaf sheath, under fruit sepals, etc. The female individuals of various mealybug species are found to release pheromone through abdominal scent glands to attract the males for mating, and this phenomenon has been exploited to develop sex pheromones for monitoring and control. Herbivore-induced plant volatiles (HIPVs) play a significant role in attracting natural enemies of mealybug, and the same could be exploited for conservative biological control. Mealybugs are also successfully used as biocontrol agents for controlling weeds.

This chapter provides a holistic description of biological attributes and life cycle details of key mealybug pests, their taxonomy and phylogeny, physiology of mealybug feeding, an overview of different strategies available for controlling mealybug pests, biological control of mealybugs and use of semiochemicals in the management of mealybugs.

Keywords

Mealybugs · Crops · Control · Semiochemicals · Invasive pests

5.1 Introduction

Mealybugs (Hemiptera: Coccoidea: Pseudococcidae) are soft-bodied sap-sucking insect pests catalogued under scale insects (Hemiptera: Coccoidea) with the worldwide distribution of about 2000 described species under 290 genera (Ben-Dov 2006). They derive their common name from the mealy wax secretion which covers their body. A detailed phylogenetic analysis supported the existence of three subfamilies, viz. Pseudococcinae, Rhizococcinae and Phenacoccinae, and within the Pseudococcinae, three tribes, Pseudococcini, Planococcini and Trabutinini, have been recognized (Downie and Gullan 2004). Based on integrated molecular and morphological data, this subfamily-level classification of mealybugs has been reinstated (Hardy et al. 2008).

Mealybugs are serious pests of agro-horti cropping systems the world over with as many as 160 mealybug species being labelled as serious crop pests. These species most frequently originate from the Palearctic region (29%), followed by the Nearctic (17%), Neotropical (16%), Oriental (15%), Afrotropical (12%) and Australasian (11%) regions. About one-quarter of them are polyphagous, while 20% have been reported as pests of grasses (like rice, wheat), and about considerable extent of them are reported as pest of horticultural and plantation crops. Mealybugs are often considered as invasive pests if we take into account the recent introductions of global invasive pests worldwide owing to their polyphagous status. Sometimes, a few narrow host-range plant mealybugs have also become invasive like cassava mealybug *Phenacoccus manihoti* (Zeddies et al. 2001).

Some of the notorious species belonging to the genera *Pseudococcus* and *Planococcus* spread between the continents propelled by international trade and anthropological interventions. Mealybugs belonging to *Pseudoccus* genus such as *Pseudococcus calceolariae* (Maskell), *Ps. longispinus* (Targioni-Tozzetti) and *Ps. viburni* (Signoret) are serious pests of apple, grapes and pear in New Zealand (Charles 1993), whereas they have been reported as pests of citrus and other subtropical fruit plants in the Mediterranean region (Franco et al. 2004). The citrus mealybug *Ps. cryptus* Hempel is a major pest of coffee roots in Asia and South America, while it attacks citrus in the east Mediterranean region (Blumberg et al. 1999). Two species of *Planococcus*, viz. *Pl. ficus* (Signoret) and *Pl. citri* (Risso), have been considered as global pests on grapevine and damage mainly vineyards (Zada et al. 2003), whereas the citrus mealybug *Pl. citri* attacks primarily subtropical fruit trees and indoor ornamental plants in the Mediterranean region (Franco et al. 2004). Polyphagous mealybugs ever remain a major threat as they could adopt to newer host plants in new environments very easily. For example, the pink hibiscus mealybug, *Maconellicoccus hirsutus* (Green), a minor pest in Southern Asia, has become a major pest of crop plants in the USA, because of its polyphagous nature, feeding on wide variety of ornamentals, fruit and vegetable crops. Initially, it was reported as a pest in Hawaii during 1984, and within a few years, it has established throughout the Caribbean islands stretching up to southern California (1999) and Florida (2002). The economic damage of hibiscus mealybug in the USA has been computed to be $750 million per year (Hall et al. 2008). Two solanum mealybug species, viz. *Phenacoccus solenopsis* Ferris and *Ph. solani* Tinsley, cause extensive damage to green pepper in Israel, while it is a serious pest of cotton in India and Pakistan (Hodgson et al. 2008). Some of the common mealybug pests are depicted in Plate 5.1.

5.2 Taxonomy and Phylogeny of Mealybugs

Extensive alpha-level taxonomic studies have led to the identification of more than 2000 species, but their relationships at suprageneric level are yet to be studied. Five subfamily names have been recognized by mealybug taxonomists: Pseudococcinae, Phenacoccinae (Sulc), Rhizoecinae (Williams), Trabutininae (Silvestri) and Sphaerococcinae (Cockerell). Several combinations of these names have been applied for classification and taxonomy of mealybugs. However, these approaches remain inadequate in terms of their generic composition or diagnostic morphology in distinguishing different mealybug groups. Understanding the phylogenetic relations between mealybug genera or species would address the concerns with regard to

A. Tailed mealybug, *Ferrisia virgata*

B. Papaya mealybug, *Paracoccus marginatus*

C. Solenopsis mealybug, *Phenacoccus solenopsis*

D. Pink mealybug, *Maconellicoccus hirsutus*

Plate 5.1 Some common mealybug pests of crops

major mealybug clades. Downie and Gullan (2004) by use of three nuclear gene-based molecular phylogeny proposed a scheme for identification of mealybugs at subfamily-level classification, and accordingly, three subfamilies have been categorized, Rhizoecinae, Phenacoccinae and Pseudococcinae, and three tribes, Pseudococcini, Trabutinini and Planococcini, have been recognized within the Pseudococcinae.

Phenacoccus solenopsis was initially described by Tinsley in 1898 from specimens infesting the plants such as *Boerhavia spicata* and *Kallstroemia californica*. Later, Tinsley (1898) provided a brief description of the adult female of *Ph. solenopsis*. Another mealybug from this genera, *Ph. cevalliae*, infesting *Cevallia sinuata* was described by Cockerell (1902). Subsequently, this species name synonymized with *Ph. solenopsis* (Fernald 1903; Ferris 1950). Ferris (1950) and several authors have redescribed the adult female of this species. Recently, Ben-Dov (2006) and Hodgson et al. (2008) documented the detailed taxonomy of this species.

Miller and Kosztarab (1979) recognized morphological variations within *Ph. solenopsis* and observed that it might consist of at least two sexual species. The presence of morphological variations among specimens of *Ph. solenopsis* in different regions of India often led to misidentification of the mealybug species (Asha and Ramamurthy 2008). Recently, based on morphological features, the specimens collected from cotton (*Gossypium* spp.) have been renamed as *Ph. gossypiphilous* (Ghulam Abbas et al. 2005; Ghulam Abbas et al. 2008, 2009). However, because no type specimens were named, Hodgson et al. (2008) considered the name to be a *nomen nudum*. Also, Hodgson et al. (2008) based on the comprehensive morphological data concluded that specimens from the Indian subcontinent were not significantly different from those of the neotropics, and he synonymized *Ph. gossypiphilous* with *Ph. solenopsis*.

Paracoccus marginatus was described by Williams and de Willink in 1992, and the identity of the species is clear. There are no scientific synonyms.

Key Diagnostics Features for Identification of Important Mealybug Species

Striped Mealybug (*Ferrisia virgata*): The body colour is grey with waxy filamentous coating all over the body and with the presence of two long tail-like structures. Two dark stripes are present on the dorsum, and body fluid is light colour. This species produces egg mass or ovisac.

Long-tailed Mealybug (*Pseudococcus longispinus*): Anal wax filaments are nearly as long as that of the body and appear as long tails; second to the last abdominal segments also has a pair of wax filaments; a dorsal median stripe is noticed on the abdomen.

Citrus Mealybug (*Planococcus citri*): Body colour is yellow to pink and covered with medium-sized slightly curved waxy filaments and is not pyramid shaped. One dorsal median stripe is present on the back in adults, and body fluid is clear. Anal filaments are less than one-eighth the length of the body. This species produces an egg mass or ovisac which is irregular and remains under the body of female.

Solanum Mealybug (*Phenacoccus solani*): The body is covered with very short waxy filaments. Long tails and stripes on the body are absent. This species does not produce an egg mass or ovisac.

Pink Sugarcane Mealybug (*Saccharicoccus sacchari*): The mealybug is light pink in colour and occurs underneath leaf sheaths on sugarcane. Adult females are provided with mobile legs.

Pineapple Mealybug (*Dysmicoccus brevipes*): This species is associated with roots of host plants. Body colour is light pink to grey and is provided with 17 pair of wax filaments.

Pink Mealybug (*Maconellicoccus hirsutus*): Mealybug colonies contain immature as well as mature females. Larger mealybugs are darker in colour and covered with significantly more white waxy material. There are no stripes on the body, and this mealybug produces an egg mass or ovisac which is irregular and remains beneath the body. When squashed, a pink to red fluid is observed.

Solenopsis Mealybug (*Phenacoccus solenopsis*): The insect is provided with short- to medium-sized waxy filaments around the body, anal filaments about one-fourth the length of the body and two dark stripes on either side of the middle "ridge" of the body. Long glassy rods are present on the back. This species produces an egg mass or ovisac.

Mango Mealybug (*Drosicha mangiferae*): The females can be identified by their flat body, covered with white mealy powder.

5.3 Life History

Life cycles differ a little from species to species. Adult mealybug females of most of the species lay about 100–200 eggs for period of 10–20 days. Egg sacs of mealybugs may be attached to leaves, crowns, twigs, barks, fruit, etc. However, the long-tailed mealybug produces eggs that reside inside the female until it hatches. The crawlers (newly hatched nymphs) are yellow or orange or at times pink in colour. The crawlers lack wax, and shortly after they settle down to feed, they begin to excrete a waxy coating. Though adults and older nymphs have legs and are able to move, they are not going very far or very quickly. Nymphs moult many times before they become adults. Mealybugs have about two to six generations a year, depending on the species and the climate. Mealybugs can overwinter in crevices of tree barks as eggs or as first instar nymphs on deciduous plants, grapevines. Occasionally, mealybugs are mistakenly identified with other pests like woolly aphids, cotton cushion scale and even some whiteflies and small scales which are secreting honeydew and waxy coatings and are associated with black sooty mould. The biology of mealybugs varies slightly between different species. The biology of some important mealybug pests is furnished in Table 5.1**.**

5.4 Feeding Physiology

Mealybugs are sap-sucking insects feeding on phloem of the plants. As they feed, mealybugs produce a sugary excretion (honeydew) that supports the growth of a sooty mould. They suck up sap by inserting their stylets from either mesophyll or phloem or both the plant tissues. Males stop feeding towards the end of the second instar. Usually, by secretion of solidified saliva forming a salivary sheath all over the stylets, the stylets are inserted into the plant tissues. Mealybug's diet is principally made up of carbohydrates and small amounts of nitrogenous compounds and free amino acids (Gullan and Martin 2003). So, food digestion is hardly required except for sucrose hydrolysis. Nevertheless, the processing of organic compounds ingested from the phloem sap occurs at filter chamber, a specific part of the digestive system that allows the clear passage of water from the anterior midgut to the Malpighian tubules, thus absorbing the food in the midgut (Terra and Ferreira 2003). After

Table 5.1 Biology of important mealybug species

S. no.	Mealybug species	Biology
1.	*Ferrisia virgata* (Ckll.)	Reproduction is sexual and parthenogenetic, while the latter is more prevalent. The longevity of the female is around 50 days, and fecundity varies from 300 to 700 eggs/female. The eggs are laid in groups, rarely alone and usually hidden beneath the body. The texture of the eggs is round and buff to light yellow. The mean duration of first instar is 6.7 days, and cocoon formation takes about 4.4 days from the first moult. In males, the total nymphal period is around 20 days, and in females, it is 43.2–92.6 days. The adult males live 1–3 days. The sex ratio (male/female) is 1:1.87. Adult females are wingless with two long conspicuous waxy filaments at the rear end, and a lot of waxy or shiny hair coated in white waxy powder covers the body..
2.	*Geococcus citrinus* Kuwana	Reproduce though bisexual and the females produces about 113–188 eggs in chains or masses that are translucent, elongate oval in shape and pearly white. The average egg period is about 12.20 days. There are three nymphal instars that lasts 7.3, 5.6 and 5.8 days, respectively. The pupal duration in male lasts 5 days. The male and female live for around 5 days and 12.78 days, respectively. Both adults and nymphs do not produce honeydew. The total life cycle is about 28.8 days..
3.	*Maconellicoccus hirsutus* (Green)	Parthenogenesis is the primary mode of reproduction; however, sexual reproduction has also been reported. The freshly deposited eggs are transparent and light orange or yellowish. The period of incubation ranges from 5 to 7 days. The first instar nymphs are typically yellow to orange with reddish eyes and last 7–9 days. The second instar nymph continues to develop for 6–8 days; at the end of the second instar, the female nymphs moult as normal, like the earlier instars, but the males secrete a cottony puparia around their body. The last instar of the female nymph period is 8–10 days. Third instar male nymphs are known by denudation of the puparia, which is characterized by the appearance of two tiny wing buds and lasts for 1–2 days. The last instar of the male nymph lasts approximately

(continued)

Table 5.1 (continued)

S. no.	Mealybug species	Biology
		5–7 days and is featured by well-developed wing pads. Development time, from egg to adult in female and male, is 30.3 and 28.7 days, respectively. The pre-oviposition period, oviposition period and fecundity ranges from 6 to 7 days, 7 to 9 days and 426 to 573 eggs. The longevity of adult females and males ranges from 13 to 16 days and 3 to 5 days..
4.	*Paracoccus marginatus* Williams and Granara de Willink	They are sexually and parthenogenetically reproduced. One female lays roughly 230–400 ovisac eggs. The ovisac is three or four times the length of the body and is fully covered in white wax. Egg laying generally takes place over a span of 1–2 weeks. The egg hatching takes place within 10 days and is greenish-yellow. The male nymphs have four instars, and the period of the first, second, third and fourth instars in the male nymph was 6.5, 6.6, 2.4 and 41 days, respectively. The fourth instar is produced in a cocoon and is known as a pupa. Adult males seem to be pink in colour, particularly during the prepupal and pupal periods, but in the first and second instars, they appear yellow. The duration of the female nymph first, second and third instar was 6.5, 5.5 and 5.2 days, respectively. Females take longer periods of development (27–30 days) than males (24–26 days). The longevity of males and females is 2.3 and 21.2 days, respectively..
5.	*Phenacoccus manihoti* Matile-Ferrero	Reproduction is by parthenogenetic thelytokous forms. Female lays 700 eggs that are wrapped in felt wax thread ovisacs. The mean female longevity is 34.3 days. The average duration of the egg, first instar, second to fourth instar and adult is around 8.0, 4.5, 4.1, 4.2, 5.2 and 20.2 days. The mean generation duration is 28.48 days..
6.	*Phenacoccus solenopsis* (Tinsley)	Reproduced by ovoviviparous means; the adult female is capable of reproduction only if she mates with a male. The female lays around 500 eggs in the ovisac; they are tiny, light yellow in colour and oval in shape. It keeps its eggs in the body until they become ready for hatching, and the incubation period is 6.6 days. The male nymphs moult four times, while the females moult three times.

(continued)

Table 5.1 (continued)

S. no.	Mealybug species	Biology
		The mean duration of the first, second and third instars is 4.8, 5.6 and 6.4 days, respectively, with the total nymphal period of the females being 16.8 days. The adult female is wingless and medium to dark yellow in colour and oblong in shape. The duration of pre-oviposition, oviposition and post-oviposition is reported as 4.3, 8.0 and 2.7 days, respectively. A female adult survives 15.5 days and has a lifespan of about 31 days. Three pairs of dark spots on the abdomen and one pair on the thorax and containing two longitudinal stripes. The males construct the puparia at the end of the second nymphal instar. The duration of the pupal period ranges from 6 to 7 days..
7.	*Rastrococcus invadens* Williams	The male and female nymphs complete their development in 38.16 and 34.67 days. The female/male ratio varies from 1:2.13 to 1:3.3. The highest fecundity, pre-oviposition period and oviposition period are 145–175 nymphs, 20–29 days, and 34–45 days, respectively, during winter..
8.	*Planococcus citri* (Risso)	In the ovisac, the female lays 300–800 yellowish-white eggs, and these hatches within 6–10 days. The male nymphs have three instars and a prepupal stage, while the female nymphs have four instars. Cottony cocoon can be produced only by males. The male adult is winged with long antennas, greyish in colour and midge-like. Total life cycle is finished within 30–35 days. The adult female body is covered in white wax and features a distinctive narrow grey line along the dorsal edge. The females, depending on the host plant, live for up to 30–40 days. The males have two long white wax threads that extend backwards. .
9.	*Pseudococcus* sp.	Oviparous reproduction and produces 650 to 900 yellowish eggs that last 5–8 days. The nymphs usually remain in the ovisac for 1 or 2 days after hatching. The crawlers (first instar), once they start feeding, produce a white waxy material that coats their body and secretes around 34 leg-like filaments around the body perimeter. The female has three nymphal instars; the female and male nymphs are indistinguishable in the first instar, but it is possible to distinguish between the sexes by the end of the second instar.

(continued)

Table 5.1 (continued)

S. no.	Mealybug species	Biology
		Female nymphs are identical to that of adult female, except the latter which are bigger. The period of female nymphs is between 18 and 21 days. The male produces pupa (cocoon) over their bodies at the end of the second instar. The developmental period of male (nymphal and pupal stages) is between 18 and 20 days. The adult male does not feed and soon dies after maturing. The male and female finish their development in 23–26 days and 25–29 days, respectively..
10.	*Saccharicoccus sacchari* (Cockerell)	Reproduced through parthenogenetic form. A single female is able of lay nearly 1000 eggs that hatch within 3–4.15 h with a duration of pre-oviposition of 13.83 days. Sometimes, only orange tiny crawlers can be found moving from below the female. The first instar is transparent, tiny and pink and more active, and they lasts for 5.3 days. The second and third instars are finished in 4.83 days and 17.2 days.

digestion and assimilation, the residue of the ingested phloem sap is released from the anus as a sugar-rich material (honeydew). This can consume up to 90% of swallowed sugars (Mittler and Douglas 2003).

Mealybugs predominantly show a phloem-feeding behaviour with extracellular or intercellular stylet pathways. However, these bugs display atypical extracellular penetration with one branch of the salivary sheath containing the actual stylets causing little damage to the phloem vessel. The saliva contains the required enzymes degrading the pectin and cellulose, the principal constituents of plant cell wall. The role of the saliva in sucking insects is of paramount importance as it is thought to contain essential digestive enzymes to help the stylets penetrate the plant tissues via formation of a kind of tunnel.

Although the feeding of mealybugs through insertion of stylets induces little mechanical damage to host tissues, they may be more toxic to the plant due to the presence of pectic enzymes and other salivary proteins. The characteristic leaf curling associated with mealybug infestation is caused due to calcium extraction linked to degradation of pectin of the leaves by salivary pectin esterase. The electrical penetration graph (EPG) studies have shown that mealybugs can ingest the intracellular calcium involved in cell wall rigidity. However, the intact nature of the cellulose layer in plant cell wall after stylet penetration of mealybugs suggests the absence of cellulases in the saliva of mealybugs. It is to be noted that salivary enzymes are altogether absent in other Sternorrhyncha and in a number of aphid species too. The callus deposits are noticed in mealybug damage in certain resistant host plants. Callus formation can be attributed to the resistance mechanisms of host plants to mealybug infestation. Callus deposition may not be regarded as an effective plant defence mechanism, and it can rather be a non-specific plant defence mechanism to counter the stylet insertion by mealybug.

Sucking pests including mealybugs have physiological association with endosymbionts to supplement the suboptimal nutrition they derive from phloem sap. These endosymbionts are considered important to their hosts' nitrogen and sterol requirements and may play a major role in resisting entomopathogens or detoxifying secondary plant metabolites (Gullan and Kosztarab 1997; Baumann 2005). Mealybugs harbour prokaryotic endosymbionts referred to as P-endosymbionts (Thao et al. 2002; Kono et al. 2008). The P-endosymbionts of the mealybugs have the peculiar feature of housing secondary endosymbionts or S-endosymbionts within their cytoplasm (von Dohlen et al. 2001). The endosymbionts are unrecognizable in the adult males as the bacteriome gradually degenerates in the prepupa and pupa during development (Kono et al. 2008).

5.5 Nature of Damage

Mealybug feeding induces symptoms such as chlorosis, defoliation, stunting and wilting. Since mealybugs feed on sugary plant juices, their waste (honeydew) contains large sugar content. Honeydew can support the development of sooty mould fungi that are not injurious to the plant tissues, but can inhibit photosynthesis in leaves if they are found in high enough concentrations. Mealybugs primarily suck the sap from plant phloem, decreasing plant vigour, and excrete sticky sap and wax that decrease the quality of plant and fruit, especially when the sooty mould grows. High populations that feed on leaves or stems can delay plant growth causing leaf fall, but without significant damage, healthy plants can withstand low populations. Ground mealybugs feed on roots of the plants causing decay of plants.

The mealybugs affect many types of perennial plants. Among the fruit crops, mealybug damage is greatly pronounced in citrus trees followed by pome and stone fruit trees. Mealybugs can attack both roots and aboveground parts of grapevine. Some woody ornamental plants, including cactus, figs, hibiscus, jasmine and oleander, are commonly infested by different mealybug pests. The cypress mealybug can also be an important pest on juniper species and cedar. Indoor plants or plants in greenhouses are particularly susceptible because mild temperatures attract mealybug colonies throughout the year. Of many houseplants, aglaonema, cactus, coleus, dracaena, ficus, ferns, poinsettia, philodendron, orchids, palms, schefflera and a variety of herbs such as rosemary and sage are attacked by mealybugs above ground. Infestations of ground mealybug are reported most often with gardenias and African violet.

Across the world, mealybugs trigger a series of economic damage on crop plants. The damage from the mealybugs is linked to their sap-sucking behaviour. The host plant often has severe infestations causing stunting or death. Mealybugs sometimes have toxins; the pink mealybug, *M. hirsutus*, reportedly found in California's

Imperial County, has saliva that is particularly toxic to plants. Several mealybugs can transmit viruses infesting a wide range of crop plants.

5.5.1 Direct Damage

Mealybugs are sap-sucking pests primarily feeding on phloem using long, slender mouthparts. Many mealybugs may feed on the leaves, trunk, fruits or flowers, stem and roots. Nonetheless, the extent of damage done by each species depends on several factors such as preferred feeding locations, population abundance (e.g., female fecundity and number of generations per year) and temperature tolerances. The honeydew excreted by these insects accumulates on plant parts. The fact that honeydew is a substratum for the production of sooty fungi which may cause more plant damage has been known for a long time. The sooty moulds can inflict cosmetic damage to the fruit cluster and decrease marketability for table fruit farmers. The infection of a limited mealybug population and of the subsequent honeydew droplets will not lead to economic damage in most dried fruits, juices and wine grapes. The honeydew deposits are washed off by rain or dried up in warm temperatures. Heavy infestation of mealybugs leads to the covering up of host plants with a hard wax-like coating on the infested plants. The bark of the plant appears water-sprinkled with a large amount of honeydew. Mealybug damage can contribute to defoliation and cause vine death following recurrent annual infestations. The plants would have modified morpho-histological features owing to the infestation of mealybugs; linear stem growth and petiole development are halted morphologically, resulting in decreased thickness of stems. The lamina of the leaf is significantly reduced and distorted. The cells would have histologically been enlarged, and the lignifications would be minimized. The infestation of *R. invadens* induced considerable decrease in mango fruit weight and sizes, as well as a significant reduction in the ash, raw fibre and sugar loss in Sri Lanka (Tobih et al. 2002).

5.5.2 Indirect Damage

Virus transmission is a primary problem, instead of feeding or contamination due to mealybug feeding in certain crops. Many mealybug species are vectors of virus diseases in crops like black pepper, banana, grapevine, pineapple, cocoa and sugarcane. Severe infestations by *Pl. ficus* can cause cluster infestation, defoliation and redness. Leaf chlorosis and phloem disruptions arise. Infection by grapevine leaf roll virus has an effect on growth and development of berries and qualitative damage on sugar content, colour changes and increased acidity of fruit juices. In some areas, toxins from mealybug are rather important. In the Hawaiian islands, the pineapple mealybug *D. brevipes* vectoring the pineapple wilt causes a serious economic loss on pineapple. Cacao swollen shoot, transmitted by mealybug vectors such as *F. virgata* and *Pl. njalensis*, causes considerable economic damage in cacao plantations.

Virus diseases transmitted by mealybug vectors follow a semi-persistent mode of transmission. Virus transmission by mealybug vectors is found to have low inoculation rate but significantly higher transmission rate. Badnaviruses such as PYMV, TaBV, BSV and CSSV are said to be spread by various species of mealybug. Citrus mealybug, *Pl. citri*, and sugarcane mealybug, *S. sacchari*, colonizing bananas spread the banana streak viruses (BSVs (Lockhart et al. 1992). Sugarcane bacilliform virus (SCBV) transmitted by *S. sacchari* from sugarcane to banana (Lockhart and Olszewski 1993) is serologically similar to BSVs (Lockhart and Autrey 1988). The striped mealybug, *F. virgata*, vectors banana streak Mysore virus (BSMYV) on banana plants (Selvarajan et al. 2006). Three mealybug species (*Pl. citri*, *Ps. ficus* and *D. brevipes*) are transmitting episomal banana streak OL virus (BSOLV) to banana (*Musa* sp.) cv. Williams (Meyer et al., 2008). The citrus mealybug, *Pl. citri*, transmits the banana streak GF virus (BSGFV) and episomal BSOLV. The pineapple mealybug, *D. brevipes*, also transmits episomal BSVs. Cocoa swollen shoot virus (CSSV), a badnavirus, is spread within the Coccoidea (Roivainen 1976) by about 14 species of mealybugs of the family Pseudococcidae, but the most important vectors are *Pl. njalensis* and *Pl. citri* (Dongo and Orisajo 2007). The piper yellow mottle virus (PYMV) is spread by *Pl. citri* (Lockhart et al. 1997) and also transmitted from genetically diseased black pepper to stable black pepper seedlings by *F. virgata* (Bhat et al. 2003).

Mealybugs also can vector a cluster of viruses, such as the mealybug wilt of the pineapple caused by a complex consisting of grapevine leaf roll-associated viruses and three pineapple mealybug wilt-associated viruses (PMWaV) (Sether et al. 2005). The disease of grapevine leaf roll is caused by grapevine leaf roll-associated viruses (GLRaVs). Such viruses are widespread worldwide in vineyards and are often connected with vitiviruses involved in the grapevine rugose wood complex. Ten species of mealybugs are classified as vectors for one or more of these grapevine viruses, including *Ph. aceris*, which is capable of transmitting the grapevine leaf roll-associated viruses 1 and 3 (GLRaV-1 and GLRaV-3). Vitiviruses, including grapevine virus A (GVA), grapevine virus B (GVB), grapevine virus D (GVD) and grapevine virus E (GVE), follow a semi-persistent spread on grapevine vines through many mealybug genera including *Cavariella*, *Pseudococcus*, *Phenacoccus*, *Planococcus*, *Heliococcus*, *Parthenolecanium*, *Neopulvinaria* and *Ovatus* (La Notte et al. 1997).

5.6 Economic Importance

Mealybugs are phytophagous insect pests affecting a wide range of host plants. There are about 160 species of mealybugs recognized as crop pests according to Millar et al. (2002). Mealybug as a pest, inflicts severe economic yield loss and affects quality of the produce in several crops and ornamental plants. Many large outbreaks of mealybugs have occurred over the past 30 years, troubling crop damage due to accidental introduction or invasions. An estimate observed that the cost of managing avoidable yield loss due to mealybug damage was computed to be around

$9.8 million in Georgia (USA) in the year 1996. Damage and cost of managing the pink hibiscus mealybug have recently been measured at $700 million per annum in the USA. Costs for controlling vine mealybug in vineyards of South Africa have been estimated to be around $100/ha/season. Most of the economically important mealybug species are polyphagous pests of various crops in different conditions. There are four aspects that economic damage will happen:

(i) A large population of mealybugs associate with leaf/flower/fruit/drops, deformation of flower and fruits and the discolouration of rind of the fruit, flowers, etc.
(ii) The honeydew excreted by mealybugs harbours the sooty mould fungus, and the black colouration of this sooty mould stains the fruit/flower causing a delay in the growth of the fruits and flavour. Photosynthetic ability, especially for young trees, may be negatively affected if sooty mould infection is serious.
(iii) Possibility of rejection of the consignment may be rejected in some export markets, if mealybugs are found on fruit/flower intended for exports as mealybug is a phytosanitary concern.
(iv) Mealybugs are vectors of plant virus disease causing severe yield losses. Many mealybugs are liable for the transmission of grapevine leaf roll-associated virus (GLRaV) infection, and the economic impact of grapevine leaf roll-associated virus 3 (GLRaV-3) infection has been estimated to reach more than $10,000 per hectare annually in South Africa.

Mealybug dispersal is facilitated by international trade. There are 350 mealybug species in the USA. Roughly 70% of the mealybug species are perceived to be invasive. The invasive mealybugs pose significant threats to a variety of economically important crops. In New Zealand, most of the 114 species of mealybugs are reported to be pests on native plants. Three cosmopolitan and invasive species of *Pseudococcus* constitute >99% of the mealybug fauna in orchards and vineyards (Charles 1993) and are regularly occurring as pests of horticultural crops. Although all the reported mealybug pests on the grapevine in France are native (Sforza 2008), they cause significant damage on grapevine in recent years (Streito and Martinez 2005). Although statistics are not available, there have been several reports on economic losses sustained by mealybugs on crop plants in many countries.

5.7 Host Plants

Mealybugs feed on a number of woody and herbaceous plants including families of angiosperms, gymnosperms and ferns. However, majority of the mealybug pests are pests of herbaceous plants especially belonging to composites (Asteraceae) and grasses (Poaceae) (Kosztarab and Kozar 1988; Ben-Dov 2006). As predicted, knowledge about the mealybugs host ranges is primarily obtained from economically significant species observations. Many species tend to be oligophagous or stenophagous or monophagous, some of which are polyphagous (Kosztarab and

Kozar 1988; Ben-Dov 2006). Many of the economically significant species are associated with long lists of hosts, and their success varies widely, varying from dense population development, which would eventually kill the host plant. Conditions of plant growth can have a strong impact on population success under fertilization and irrigation. Most of the mealybug species may easily be mass cultured on alternate hosts during laboratory studies, such as potato sprouts or squashes. For instance, citrus mealybug has been identified on a wide variety of host plants belonging to 70 botanical families; 60% of these are classified as non-woody plants, whereas on a global scale, the citrus mealybug is a pest of tropical and subtropical crops such as persimmon, citrus, custard apple, and banana, or it damages various types of plant species in indoor environments, especially in greenhouse. The case of *Ps. cryptus* is another example of the apparent contradiction between the long lists of host plants and the small ranges of damaged crops; although this mealybug is reported from 35 host plant families (Ben-Dov 2006), it causes damage only to citrus trees in Israel.

5.8 Mealybug "a Hard to Kill" Pest

Mealybugs are notorious pests and pose a threat to diverse cropping system across the globe. Being a quarantine pest, it poses a serious risk of introduction in new territories. Despite being a soft-bodied and delicate insect, mealybugs are difficult to be controlled for several reasons. Mealybugs are "hard to kill" insects due to its morphological, structural and behavioural adaptability (Mani and Shivaraju 2016). They have a cryptic behaviour and clumped spatial distribution, and many of the species have a tendency to aggregate and establish themselves in cracks and crevices in bark and leaf axils, under the sheath, under fruit sepals and between touching fruits or fruits and leafs (Kosztarab and Kozar 1988). This cryptic behaviour makes mealybugs practically invisible during the latent population phase and provides a spatial and temporal refuge from natural enemies and harsh weather conditions (Gutierrez et al. 2008).

Mealybugs are known for their dermal wax secretions in all life stages except in few early nymphal instars. Plant protection chemicals are of limited efficacy against mealybugs because of their concealed habitat and due to the presence of waxy coating over its body. Hydrophobic waxy covering repels hydrophilic insecticides and prevents them from entering in the body. Also, the eggs, concealed in waxy filamentous ovisac, are almost impossible to be penetrated by insecticides (Cox and Pearce 1983; Foldi 1983). The wax cover avoids water loss and prevents them from desiccation during high temperature. Overlapping generations with staggered age structure present simultaneously consisting of all the life stages (eggs, crawlers and adults) demand frequent insecticide application which ultimately leads to development of insecticide resistance. Many of the mealybug species are protected by ant guards for honeydew; in return, ants help in dispersal of mealybugs and offer protection against natural enemies. Many traditional insecticides are neither effective against mealybugs nor compatible with natural enemies, and repeated use of these insecticides will kill existing natural enemies. Certain insect growth regulators may stimulate development and reproduction of mealybugs (Cloyd 2003). Owing to all these morphological, physiological and behavioural adaptations of mealybugs, they remain well protected from high summer temperatures, natural enemies and most insecticides.

5.9 Management Measures

Modern approaches of pest control are aimed at "pest management" rather than "pest control." Best suitable, available, eco-friendly and economically viable pest management tools are integrated to achieve effective and efficient pest management. These tools include cultural, mechanical, chemical, biological and host plant resistance. Different tools in relation to mealybug management are discussed in the following section.

5.9.1 Cultural Practices

Mealybugs are most often introduced on new plants or on pots or tools into the landscapes (and particularly indoor areas). As the adult females are unable to fly and mealybugs are unable to crawl very fast, they are not dispersing rapidly in the garden themselves. Inspection of mealybugs carefully before planting some new plants and removal of the same from saplings could help prevent the spread of the mealybugs. There are a number of cultural control measures widely adopted in different regions. Sanitation of crops is helpful to reduce the population of mealybug. It is best to manually remove fluffy nests and most insects before adding the insecticides/pesticides. The weeds carrying mealybugs are to be destroyed so as to remove the spread onto cultivated plants. All crop residues and trashes are to be cleared once the crop is harvested as the leftover crop residues in the field may harbour mealybug pests which may infest the succeeding crop. The removal of bark reveals certain mortality factors to mealybugs.

Inspect mealybug-prone plant species periodically for mealybug infestation, and dispose any infested objects containing their egg sacs. Exposure to forced water sprays can be tried at regular intervals. The control of some garden mealybugs may be achieved by flushing a strong stream of water (Michelbacher et al. 1959). Avoid excessive use of nitrogenous fertilizers on crops as high nitrogen levels coupled with daily irrigation will boost tender growth of new plants as well as the production of mealybug eggs. In case of interior plants, use of only plant species not susceptible to mealybugs could prevent continuous build-up of mealybug pests. Ground mealybugs are even harder to control than over ground ones.

Physical barriers can be kept parallel to the field periphery to keep ants away from field and thereby control mealybug populations. Removal of weeds from field bunds and crop debris deters the movement of mealybugs between plantings.

Cover crops can be used by enhancing natural enemy numbers or variability to boost soil health and lower mealybug densities. Parasitoids attacking mealybugs might use some cover crop species and floral nectars to provide food for the longevity of adults. Generalist predators, such as ladybird beetles and lacewings, could also use these floral food supplies as well as herbivores as alternative prey in the cover crop. Excessively strong plants may increase the populations of mealybug in two respects. It has been shown that excess nitrogen increases the abundance of mealybug females and the number of eggs in each ovisac. The enhanced vegetation associated with vigorous plant growth provides the mealybugs an improved protection by lowering the temperatures inside the crop canopy and may reduce the spreadability of foliar insecticides.

5.9.2 Physical Control

A number of physical measures are of help to contain the mealybug incidence and damage (Gould and McGuire 2000):

(i) Application of water streams to flush off mealybugs from plant surface.
(ii) Scraping and smashing the colonies with cloth.
(iii) Mealybugs can be extracted also by tubing cotton swab to a polishing alcohol or fingernail.
(iv) Discarding severely mealybug-infested plants.
(v) Hand-picking of mealybugs and crushing the same – this practice can be of use in kitchen gardens or conservatory plots for germplasm lines.
(vi) Covering of plant parts such as fruits with paper bags to avoid infestations.
(vii) Removal of loose barks from trees to minimize hiding places for mealybug colonies and swabbing of stems with a solution containing dichlorvos 76 EC at 2 ml/litre alone or in combination + fish oil rosin soap emulsion (2% concentration).
(viii) Banding of trees with mineral oil or sticky bands to prevent movement of crawlers from tree trunk to branches.
(ix) Deep ploughing and summer ploughing and destruction of field bunds to destroy the colonies of root mealybugs hiding near bushes.
(x) Application of hot water (about 50 °C for two minutes) has also been found to be effective against mealybugs such as *Planococcus citri* and *Pseudococcus odermatti*.

5.9.3 Host Plant Resistance

Development of moderately resistant/resistant varieties is another approach to pest population management in combination with biocontrol. There is not much information available on utility of resistant cultivars in the management of mealybugs in crop plants. Available literature indicates non-availability of indigenous cultivars

Table 5.2 Varieties/cultivars identified as sources of resistance or tolerance to mealybugs

Crop	Varieties/cultivars	Mealybug species	Mechanism of resistance	References
Cassava	Ganfo, Incoza and Zanaga	*Phenacoccus manihoti*	Tolerance	Le Ru and Tertuliano (1993)
Mango	Tukhmi, Anwar Ratul	*Drosicha mangiferae*	Resistance	Karar et al. (2015)
Bermuda grass	Princess, TifSport	*Antonina graminis*	Resistance	Reinert et al. (2009)
Rice	TNAU 831520, TNAU 831521, Tending, Badal 2, Rathu Heenati, Ptb 21, Sufaida 172, IR 42 and IR 72 and breeding lines IR 54751–1–19-13-17-3 and IR 54742–1–19-11-8-2	*Brevennia rehi*	Resistance	Jayarani (1992) and Backialakshmi (1994)
	TNAU 80030, TM 1087 and CO 43	*Brevennia rehi*	Tolerance	Mallikarjuna Rao (1987)

nor improved crop cultivars with high levels of resistance to mealybugs (Ayyar 1939; Alam et al. 1979; Heinrichs 1983). However, some of the recent studies have highlighted the utility of host plant resistance mechanisms in important crop varieties against mealybugs (Table 5.2).

5.9.4 Chemical Control of Mealybugs

Chemical control is still the most commonly used management practice for effective control of mealybugs although there are constraints imposed on their use in different cropping systems (Franco et al. 2009). Mealybugs have great reproductive potential; reproduction is very high during favourable weather conditions. Abundance of mealybugs has made it impossible for the natural enemies available in the field to keep these pests under control. Hence, chemical control is resorted to protect the crops form mealybug damage.

Choice of insecticides and adoption of appropriate application techniques are important for achieving effective control with insecticides against mealybug pests. When the timing of use of insecticides is planned to coincide with the nymphal instars of mealybugs, effective control can be achieved. Contact insecticides are little or no use against mealybugs owing to the fact that the waxy coating on the surface of the mealybugs poses a physical barrier which repels polar chemical insecticides preventing its penetration in the body (Walton et al. 2004; Mani and Shivaraju 2016). Systemic insecticides, which move through plant system, provide an excellent preventive control against mealybug colonization. When exposed to systemic insecticides, insects with sucking mouthparts withdraw and ingest lethal concentrations of active ingredient and would subsequently be killed. Traditionally, insecticides such as chlorpyrifos, diazinon and acephate among organophosphates and aminocarb, carbaryl and methomyl among carbamates with broad-spectrum action have been used for mealybug management (de Souza et al. 2007; Shafqat et al. 2007; Gonzalez et al. 2001). Many organophosphates such as profenofos have been found effective against cotton mealybug, *Ph. solenopsis* (Dhawan et al. 2009). Effective control can be achieved with these old insecticides by coinciding the timing of application with vulnerable young nymphal instars on the crop. However, the older group of compounds is being replaced by chemicals with better efficacy in the recent past.

5.9.4.1 Novel Insecticides

Newer insecticidal chemistries, with novel modes of action, have better efficacy against mealybug pests. New insecticides belonging to neonicotinoids, insect growth regulators and biosynthesis inhibitors gained recognitions. Neonicotinoid group of insecticides shows high effectiveness against mealybugs with negligible effect on non-targeted organisms. These compounds owing to better systemic action have the potential to replace carbamates, organophosphates or pyrethroids. Imidacloprid and thiamethoxam among neonicotinoids are found to be effective in control of grapevine mealybug, *Planococcus* sp., and other mealybug pests of horticultural crops (Ambethgar 2011). When imidacloprid wettable powder (WP) formulation was compared with soluble liquid concentrate (SL) formulation, WP formulation was observed to be safe to natural enemies (Elbert and Nauen 2004). Other insecticides in the neonicotinoid group such as acetamiprid, thiacloprid, dinotefuran and clothianidin have also been found effective against mealybugs (Daane et al. 2006; Grout and Stephen 2005; Sazo et al. 2006).

Other class of chemicals which has shown promise towards mealybug control is insect growth regulators (IGR). Juvenile hormone analogues and chitin synthesis inhibitors are considered as a suitable alternative, as they exhibit low mammalian toxicity. Buprofezin, a chitin synthesis inhibitor, is one of the most effective IGR against mealybugs (Muthukrishnan et al. 2005); however, its efficacy is limited to eggs and nymphal instars of mealybugs. Juvenile hormone mimics, pyriproxyfen and kinoprene reduce egg production in citrus mealybug (Cloyd 2003).

Recent studies show that some novel group of insecticides like tetramic acid, sulfoximine and butenolide are effective in controlling pink hibiscus mealybug, *M. hirsutus* (Ganjisaffar et al. 2019). The lipid biosynthesis inhibitor, spirotetramat (tetramic acid), had ovicidal activity and was also effective against early nymphal instars of the mealybug. Sulfoxaflor, nAChR modulator, provided a moderate control against early nymphal instars. The nAChR agonist, flupyradifurone (butenolide), caused high mortality in eggs, emerged crawlers and second instar nymphs and also affected the oviposition in adult females.

5.9.4.2 Botanical Plant Products and Other Natural Products

Botanical compounds such as neem oil, mineral oil and fish oil rosin soap are some of the plant or natural products being used for mealybug management. Prophylactic application of botanicals and natural products at crawler stage is recommended to reduce build-up of mealybug population. They are generally considered safer than the usual insecticides.

Neem has a number of biological activities such as repellency, feeding deterrency and IGR activity against target pests with non-toxic effect on naturally occurring parasitoids and predators. Neem oil in soap emulsion or 5% neem seed kernel extract (NSKE) has been found effective against mealybugs. Neem oil disrupts the growth and development of the mealybugs reducing their reproduction cycle leading to suppression of mealybug pests. Neem oil emulsions have smothering effect on mealybugs; hence, complete coverage of plant canopy is crucial to achieve better efficacy. Neem insecticides containing azadirachtin can also be sprayed as contact insecticide for mealybug suppression (Irulandi et al. 2001).

Other botanical insecticides such as pyrethrins and rotenone have been used against mealybugs with limited effectiveness. Crude extracts of pepper and eucalyptus at 3500 ppm have shown promising results and were able to cause about 90% mortality of *Pl. citri* (Ahmadi et al. 2012). Fruit extract of the plant *Balanites aegyptiaca* has been reported to reduce the population of mealybug *F. virgata* (Cockerell) after the third day of spraying (Wabale et al. 2010). Various literatures recommend the use of surfactants while application of bioinsecticides for removal of waxy coating to expose the mealybugs. Insecticidal formulations based on *Datura stramonium*, *Azadirachta indica* and *Nicotiana tabacum* along with suitable surfactants have also been found to be effective (Pascual Villalobos and Robledo 1998; Krishnan et al. 2016).

Oils have long been used for the control of scale insects but have limited success in mealybug management. Effective control of mealybugs on ornamental plants can be achieved by applications of summer oil emulsions/mineral oils at regular intervals of 1–3 weeks. Integration of narrow refined oils and oil emulsions with other contact insecticides was suggested to dissolve the insect's wax covering which improve the insecticide efficacy for controlling garden mealybugs. Insecticidal soaps are a solution of mild detergent made from petroleum products and synthetic pyrethroids. The soap penetrates the protective waxy coat created by mealybugs and kills the pests. Complete coverage of the plant parts and spraying at regular interval are important for better results. Neem oil, light mineral oils and fish oil rosin soaps can be incorporated in organic or sustainable farming programmes.

5.9.4.3 Application Techniques and Precautions

Insecticide application timing and technique are critical to control mealybugs (Daane et al. 2008a). Application of insecticides at early nymphal stage or crawler stage has been found most effective in controlling mealybugs. Early-stage nymphs with less wax cover are more susceptible than the later-stage mealybugs. Therefore, much research has been intended towards devising appropriate application method and timing of application for effective control of mealybugs.

Four principal methods are adopted for insecticide application in mealybug management: foliar application, soil drenching, chemigation with irrigation water and smearing/swabbing tree trunk with insecticides. Foliar application is the most common and widely used method to control the aboveground mealybug populations. Timing of insecticide application and stage of mealybug influence the effectiveness of foliar applications. Foliar sprays are effective when chemicals are applied when the first instar nymphs (crawlers) are on dispersal mode. Earlier studies have shown that foliar application of organophosphates such as dimethoate or trichlormetaphos is effective against mealybug pests: *Ps. maritimus* (Ehrh.) and *Pl. citri* (AliNiazee and Stafford 1972: Kurdyukov and Alan 1973). Foliar application of buprofezin is most effective and provides excellent control against smaller mealybugs undergoing moults (Muthukrishnan et al. 2005; Daane et al. 2008a). Mealybug management programmes in North America is based on foliar application of insecticides like chlorpyrifos, imidacloprid, clothianidin, acetamiprid, buprofezin and spirotetramat (Bentley et al. 2008).

Soil drenching with systemic insecticides and spot treatment with granular insecticides give better control of all mealybug stages in orchards. Drenching of chemicals and spot treatment reduce non-target effects on natural enemies. Besides, translocation of systemic insecticides offers protection for an extended time. Unlike contact insecticides, losses due to evaporation, light degradation and irrigation are minimized when systemic insecticides are used. Better control of grapevine mealybugs in India was reported by application of imidacloprid at the base of the trunk by soil drenching (Mani and Shivaraju 2016). Soil drenching with chlorpyrifos 20 EC at 2.5 ml per litre of water in the root zone helps to reduce the root mealybug population in banana. Soil application of granular insecticides, namely, phorate, aldicarb, carbofuran and fipronil, has been recommended for control of mealybugs.

Application of insecticides through irrigation water (referred to as chemigation) has been found to be effective and safe to the environment. Utilizing drip irrigation system for delivering insecticides saves water and efficient delivery of insecticides for many horticultural crops. Imidacloprid and thiamethoxam having systemic transluminar action have been used in several countries, and excellent control of mealybug has been obtained for a longer time. It has been reported that application of imidacloprid through drip irrigation provides up to 90%–93% reduction in mealybug population (Daane et al. 2008b). Swabbing trunk with chlorpyrifos 20 EC at 2 mL/l in combination with fish oil rosin soap at 2 g/litre has provided better protection from grapevine mealybugs in India (Mani and Shivaraju 2016).

5.9.5 Semiochemicals

The use of chemicals which are used by insects for communication, commonly called as semiochemicals, is considered to be one of the promising tools in insect pest management. Thorough investigations have been made to understand the mode of insect interaction with its environment, especially host plants and natural enemies. The insect communicates with other individuals of the same species (intraspecific) or with individuals of other species (interspecific) mostly by using semiochemicals. The intraspecific communication is mainly facilitated by pheromones (Karlson and Luscher 1959) which are either released by males or females or both or perceived by the opposite sex of the same species and brings behavioural changes in receiving species. On the other hand, interspecific communication is governed by set of chemicals called as allelochemicals (Whitacker and Feeny 1971). With mealybugs being very small-sized insect, deciphering the role of semiochemicals is somewhat a tedious task compared to other insects belonging to Lepidoptera and Coleoptera.

5.9.5.1 Pheromones

The female individuals of various mealybug species are found to release pheromone through abdominal glands to attract the males for mating, and this phenomenon is used to develop strategies to monitor the mealybug pest. Besides abdominal glands, translucent pores on metathoracic legs have been found to be pheromone-releasing site in *Ps. viburni* (Signoret) and *Pl. ficus* (Waterworth et al. 2012). Isolation of pheromone from female is one of the important and crucial steps for accurate identification of pheromone components. Although females of mealybugs release pheromone for weeks, it lasts only till mating; hence, timing of pheromone collection plays a crucial role. It is also important to maintain female population separately in virgin state, while volatile collection due to attention must be given to avoid the interference of host plant volatiles. Because of small size of female individuals, very minute quantity of pheromone can be obtained from single female; hence, maintaining large colony is also a prerequisite. To get rid of these difficulties, most reports suggest the rearing of mealybugs on potato. Pure culture of female individuals is achieved by manually removing male individuals (Negishi et al. 1980) or by using insect growth-regulating hormones (Moreno et al. 1976). Headspace sampling technique with Porapak Q or other adsorbent (Tenax, activated charcoal, etc.) is mostly used to collect the female-released volatiles. Bierl-Leonhardt et al. (1980) were the first to identify pheromone from mealybug. They identified pheromone component for the Comstock mealybug, *Ps. comstocki* (Kuwana), which is a pest of citrus, pomegranates and other fruits in California and other parts of the world, as 2, 6-dimethyl-1, 5-heptadien-3-ol acetate. The pheromone has shown attractancy at concentration as low as 0.2 μg per trap. During the same time, Negishi et al. (1980) also reported the same pheromone component for Comstock mealybug and found potent attractancy in laboratory as well as field bioassays.

Bierl-Leonhardt et al. (1981) isolated and identified (1R,3R)-cis-2,2-Dimethyl-3-isopropenyl-cyclobutanemethanol acetate as sex pheromone for citrus mealybug, *Pl. citri*. Subsequently, Rotundo and Tremblay (1982) also confirmed the attractancy of males to synthetic pheromone lures. Pheromone from another species from the same genus (*Planococcus*) was identified as blend of (S)-5-Methyl-2-(prop-1-en-2-yl)-hex-4-enyl 3-methyl-2-butenoate and (S)-5-Methyl-2-(prop-1-en-2-yl)-hex-4-en-1-ol from females of vine mealybug, *Pl. ficus* (Hinkens et al. 2001; Zada et al. 2001). Further, Millar et al. (2005) demonstrated the attractiveness of lure loaded with 100 μg of (S)-5-Methyl-2-(prop-1-en-2-yl)-hex-4-enyl 3-methyl-2-butenoate in the field. In California vineyards, Walton et al. (2006) successfully demonstrated mating disruption by using sprayable, microencapsulated formulation of lavandulyl senecioate ((S)-5-Methyl-2-(prop-1-en-2-yl)-hex-4-enyl 3-methyl-2-butenoate). Mating disruption by application of pheromone at 93.8 g active ingredient per hectare through reservoir pheromone dispenser was found successful in vineyards at Sardinia, Italy (Cocco et al. 2014).

(E)-2-Isopropyl-5-methyl-2,4-hexadienyl acetate as a pheromone component was identified from virgin females of *Pl. minor* (Maskell), also known by the common names passion vine mealybug, Pacific mealybug or guava mealybug. It was also confirmed that (Z)-isomer of this compound is a strong antagonistic (Ho et al. 2007). The pheromone was identified by collecting headspace volatiles for first 4 h of photophase using Porapak Q as adsorbent and pentane as an eluent. Considering the requirement of isomeric purity in pheromone compound and cost, Millar (2008) suggested a three-step, stereospecific synthesis of the pheromone. The lure prepared from synthetic pheromone of *Pl. minor* proved to be effective tool for monitoring its incidence in the USA. It was further confirmed that pheromone compound is species specific and attracts only *Pl. minor* males which are morphologically very similar to *Pl. citri* (Roda et al. 2012).

A sex pheromone from Japanese mealybug, *Pl. kraunhiae*, pest of fruit crops including pear, citrus, grape and persimmon (Kawai 1980), was collected through headspace air sampling using Tenax TA, 20–35 mesh as adsorbent and hexane as eluent. The extract was further fractionated through column chromatography, and active pheromone component was identified as 2-isopropyliden-5-methyl-4-hexen-1-yl butyrate through GC-MS and NMR (Sugie et al. 2008). Tabata (2013) proposed a new and convenient route for its synthesis using commercially available 2-isopropenyl-5-methyl-4-hexen-1-ol (lavandulol), whereas Tabata et al. (2015) proposed its synthesis from natural lavandulol without isolating it from lavender essential oil.

Pheromone of pink mealybug, *M. hirsutus*, was identified as two-component blend, viz. (R)-2-isopropenyl-5-methyl-4-hexenyl (S)-2-methylbutanoate and [(R)-2,2-dimethyl-3-(1-methylethylidene)cyclobutyl]methyl (S)-2-methylbutanoate, and lure containing 1:5 mixture was found to be a potent attractant for males (Zhang et al. 2004). Further, Zhang and Amalin (2005) evaluated this blend in the field at Key Biscayne, Florida, the USA, and found that concentration of lure as low as 0.1 μg was attractive to males in sticky traps, but significantly higher trap catches were observed in traps with 1 or 10 μg blend. Trap design and size also influence the effectiveness and efficiency in trapping male individuals. Jackson trap was found to be most suitable for monitoring *M. hirsutus* which trapped more males per square centimetre compared to different other traps, viz. green Delta, Pherocon IIB, Pherocon V and Storgard Thinline traps (Vitullo et al. 2014).

Millar et al. (2005) isolated and identified a highly irregular terpenoid structure, (1R*,2R*,3S*)-(2,3,4,4-tetramethylcyclopentyl)methyl acetate in which the two isoprene units forming the carbon skeleton are joined by 2′-2 and 3′-4 connections, as a pheromone of obscure mealybug, *Ps. viburni.* Field efficacy of this pheromone compound in apple orchards using red delta trap and rubber septum impregnated with 4 μg of synthetic sex pheromone showed potent attractivity to males and also to

Table 5.3 List of pheromone components of different mealybug species

S. no.	Species	Pheromone component	References
1.	Grape mealybug, *Pseudococcus maritimus*	(R∗,R∗)-trans-(3,4,5,5-Tetramethylcyclopent-2-en-1-yl)methyl 2-methylpropanoate (trans-α-Necrodyl isobutyrate)	Figadere et al. (2007)
2.	Long-tailed mealybug, *Pseudococcus longispinus*	2-(1,5,5-Trimethylcyclopent-2-en-1-yl)ethyl acetate	Millar et al. (2009)
3.	Citrophilous mealybug, *Pseudococcus calceolariae*	Major component: [2,2-dimethyl-3-(2-methylprop-1-enyl)cyclopropyl]methyl 2-acetoxy-3-methylbutanoate (chrysanthemyl 2-acetoxy-3-methylbutanoate) Minor components: 1)[2,2-dimethyl-3-(2-methylprop-1-enyl)cyclopropyl]methanol (chrysanthemol) and 2) [2,2-dimethyl-3-(2-methylprop-1-enyl) cyclopropyl]methyl 2-hydroxy-3-methylbutanoate (chrysanthemyl 2-hydroxy-3-methylbutanoate)	El-Sayed et al. (2010)
		(1R,3R)-[2,2-Dimethyl-3-(2-methylprop-1-enyl)cyclopropyl]methyl (R)-2-acetoxy-3-methylbutanoate.	Unelius et al. (2011)
4.	*Pseudococcus cryptus*	3-Isopropenyl-2,2-dimethylcyclobutylmethyl 3-methyl-3-butenoate	Arai et al. (2003)
5.	Madeira mealybug, *Phenacoccus madeirensis*	Trans-(1R,3R)-chrysanthemyl (R)-2-methylbutanoate and (R)-lavandulyl (R)-2-methylbutanoate (ratio of 3:1)	Ho et al. (2009)
6.	Cotton mealybug, *Phenacoccus solenopsis*	(2,2-Dimethyl-3-isopropylidenecyclobutyl)-methyl 3-methylbut-2-enoate	Tabata and Ichiki (2016)
7.	Spherical mealybug, *Nipaecoccus viridis*	2,2,3,4-Tetramethyl-3-cyclopentene-1-methanol (γ-necrodol) and γ-necrodyl isobutyrate	Levi-Zada et al. (2019)
8.	Striped mealybug, *Ferrisia virgata*	(1S,3R)-(−)-cis-Chrysanthemyl tiglate	Tabata and Ichiki (2017)
9.	Matsumoto mealybug, *Crisicoccus matsumotoi*	3-Methyl-3-butenyl 5-methylhexanoate	Tabata et al. (2012)

its parasitoid *Acerophagus maculipennis* (Mercet) (Hymenoptera: Encyrtidae) in New Zealand (Charles et al. 2015). The pheromone components identified for other mealybug species belonging to different genus are summarized in Table 5.3.

The pheromone traps of *Ps. maritimus* have been successfully used for monitoring of mealybug and to study its seasonal phenology in Washington's Concord grape vineyards (Bahder et al. 2013). For 30% sampling precision efficiency, four to eight traps per 12.14 ha are required, whereas to know only flight phenology, one trap per 12.14 ha was sufficient. Walton et al. (2013) also demonstrated the successful use of pheromone-baited delta traps to detect the initiation of flight activity and incidence in the grape vineyards at Southern Oregon, Columbia Basin and Willamette Valley (USA). Monitoring through pheromone-baited traps were found to be better than visual monitoring. Deployment of pheromone for monitoring flight activity and mass trapping has been successfully demonstrated for citrophilous mealybug (*Ps. calceolariae*) in citrus orchards at South Australia (Sullivan et al. 2019).

5.9.5.2 Kairomones

Not much research has been done to identify host plant volatiles which could play a role in attraction of mealybug to the host plant. But there are several reports on prey location by mealybug predators and parasitoids using volatiles emanated from infested or uninfested host plant. Herbivore-induced plant volatiles (HIPVs) are the volatiles released from host plant due to herbivory or infestation of insect pest. The role of these volatiles in attracting natural enemies of mealybug could be seen from reports of Bertschy et al. (1997) and Bertschy et al. (2001) where they demonstrated that *Apoanagyrus* (*Epidinocarsis*) *diversicornis* Howard (Hymenoptera: Encyrtidae) and *Aenasius vexans* Kerrich, the larval parasitoids of the cassava mealybug, *Ph. herreni*, showed preference towards infested plant than healthy plant. Parasitic wasps, viz., *A. coccois* and *A. vexans*, were also found using contact kairomone, O-caffeoylserine, present on the body surface of its host (cassava mealybug *Ph. herreni*) to locate the host insect (Calatayud et al. 2001). The coccinellid predator *Exochomus flaviventris* females were found to use HIPVs of the cassava mealybug *Ph. manihoti*; the predator can detect the presence of gravid females via olfaction (Le Ru and Makosso 2001). Apart from plant or prey volatiles, pheromone compounds released by mealybugs are used as cue to locate mealybug colony by its natural enemies. The pheromone of vine mealybug, *Pl. ficus* ((S)-5-Methyl-2-(prop-1-en-2-yl)-hex-4-enyl-3-methyl-2-butenoate(S-lavandulyl senecioate)), was found to be used by its parasitoid, *Anagyrus pseudococci* (Franco et al. 2008; Franco et al. 2011). Using "potato trap" technique, Mansour et al. (2010) also confirmed the role of S-lavandulyl senecioate in attraction of *A. pseudococci*, thereby enhancing the biological control of mealybug in vineyards at Sicily, Italy.

5.9.6 Biological Control of Mealybugs

Mealybugs are hard to be killed as they inhabit concealed locations and have protection offered by waxy coating to the nymphs, and adults act as physical barriers to penetration of insecticides. Even though different management strategies are prevailing, a failure of chemical insecticides in providing satisfactory control of mealybug and added advantages of biological pest suppression, viz. sustainability, less ecological foot prints/environmental friendliness and excellent success rate, call for biological management of insect pests. Biological control strategies through use of entomopathogens, predators and parasitoids provide excellent control of mealybugs on different crops. Some of the successful biocontrol agents deployed against mealybugs are depicted in Plate 5.2.

	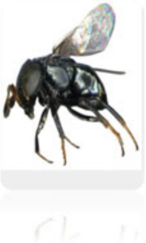
A. Cotton mealybug parasitized by *Aenasius arizonensis*	Adult parasitoid of *Aenasius arizonensis*
	
B. Papaya mealybug parasitized *by Acerophagus papayae*	Adult parasitoid of *Acerophagus papayae*
	
Predatory coccinellid , Cheilomenes sexmaculata	Predatory coccinellid , Coccinella septempunctata

Plate 5.2 Natural enemies of some mealybug pests

Globally, biological control agents have played a crucial role in suppressing the different mealybugs, viz. pink mealybug, *M. hirsutus* (Green), in the Caribbean (Kairo et al. 2000); cassava mealybug, *Ph. manihoti* Matile-Ferrero, in Africa (Herren et al. 1987); cotton mealybug, *Ph. solenopsis*, in Pakistan (Solangi et al. 2012); and citrus mealybug, *Pl. citri* (Risso), in India (Singh 2004). Biological control strategies deployed against some of the key mealybug pests are discussed hereunder.

5.9.6.1 Natural Enemies in Management of Papaya Mealybug, *Pa. marginatus*

A handful of predators and parasitoids associated with *Pa. marginatus* were recorded from different agroecosystems and emphasize the usefulness of biocontrol for successful management of these pests. A number of predators like Coccinellid beetles (ladybirds), *chrysopids* (lacewing bugs), syrphid flies and Lepidopteran predators are reported feeding on papaya mealybug populations (Table 5.4).

A number of hymenopteran parasitoids belonging to Encyrtidae family were reported as indigenous to the origin of mealybug, viz. *Anagyrus loecki*, *Pseudophycus* sp., *Acerophagus papayae*, *Pseudleptomastrix mexicana* and *Apoanagyrus californicus* augmented as classical biological control programmes (Table 5.5). Since parasitoids were proven as an effective means for mealybug control, three exotic encyrtid parasitoids, viz. *A. loecki, A. papayae and P. mexicana*, were introduced to India from USDA-Animal and Plant Health Inspection Service (APHIS), Puerto Rico, on 15 July 2010 by concerted efforts from ICAR-National Bureau of Agricultural Insect Resources (NBAIR), Bangalore, India. An average of 10.4% parasitism and 9.7% reduction in papaya mealybug population was noticed a month after release of parasitoids, and thereafter, a gradual decrease in mealybug population in response to increased parasitism percentage was noticed. Upon release, it was observed that among the two candidate parasitoids, *A. papaya* had shown higher percent parasitization (75.6–81.7%) followed by *P. mexicana*.

The fungal pathogens *Verticillium lecanii* (Zimm.), *Beauveria bassiana* (Bals.), *Paecilomyces pictus*, *Neozygites* and *Metarhizium anisopliae* (Metsch.) are known to cause 40–50% mortality of *Pa. marginatus* (Banu et al. 2010; Shylesha et al. 2011a).

5.9.6.2 Natural Enemies in the Management of *Ph. solenopsis*

Phenacoccus solenopsis was recorded as a pest of cotton in 1898 by Tinsley (1898) in New Mexico, the USA. Later on, the spread of this mealybug was noticed across the states of Mississippi, Arizona, Washington D.C., Colorado, Texas and California of the USA (McKenzie 1967). Fuchs et al. (1991) found out that this species had spread throughout several cotton (*Gossypium* spp.)-growing areas of Texas by 1988 and also recorded this pest from 29 additional plant hosts. In the recent years, a severe outbreak of cotton mealybug, *Ph. solenopsis*, was reported in the Punjab and Sindh regions of Pakistan during 2005 (Arif et al. 2009). By 2006, it had spread to a large number of cotton-growing districts severely impacting the yield of the cotton.

Table 5.4 Predators associated with mealybug, *Pa. marginatus*

Species name	Order: family	Host plants	Country	References
Brumoides suturalis (Fabricius)	Coleoptera: Coccinellidae	Cotton, papaya	India	Tanwar et al. (2010)
Cheilomenes sexmaculata (Fabricius)	Coleoptera: Coccinellidae	Papaya	India	Tanwar et al. (2010) and Jonathan et al. (2011)
Coccinella transversalis	Coleoptera: Coccinellidae	Papaya, plumeria	India	Jonathan et al. (2011)
Coccinella septempunctata Linnaeus	Coleoptera: Coccinellidae	Papaya	India	Tanwar et al. (2010)
Nephus regularis Sicard	Coleoptera: Coccinellidae	Papaya	India	Tanwar et al. (2010)
Scymnus coccivora Ayyar	Coleoptera: Coccinellidae	Papaya, plumeria	India Thailand	Tanwar et al. (2010) Saengyot and Burikam (2011)
Cryptogonus orbiculus (Gyllenhal)	Coleoptera: Coccinellidae	Papaya, plumeria	Thailand	Saengyot and Burikam (2011)
Scymnus quadrillum Motschulsky	Coleoptera: Coccinellidae	Papaya, plumeria	Thailand	Saengyot and Burikam (2011)
Cryptolaemus montrouzieri (Mulsant)	Coleoptera: Coccinellidae	Cotton, papaya	India Malaysia Hawaii	Tanwar et al. (2010) Mastoi et al. (2015) Ronald et al. (2007)
Sasajiscymnus quinquepunctatus (Weise)	Coleoptera: Coccinellidae	Papaya, plumeria	Thailand	Saengyot and Burikam (2011)
Chrysoperla pallida sp. nov.	Neuroptera: Chrysopidae	Papaya, plumeria, Cotton	India Thailand	Tanwar et al. (2010) Saengyot and Burikam (2011)
Mallada basalis (Walker)	Neuroptera: Chrysopidae	Papaya, plumeria	India Thailand	Tanwar et al. (2010) Saengyot and Burikam (2011)
Apertochrysa sp.	Neuroptera: Chrysopidae	Papaya	Malaysia	Mastoi et al. (2015)
Orius sp.	Hemiptera: Anthocoridae	Papaya	India	Tanwar et al. (2010)
Spalgis epius (Westwood)	Lepidoptera: Lycaenidae	Papaya	India Thailand	Tanwar et al. (2010) Saengyot and Burikam (2011)
Stethorus sp.	Lepidoptera: Lycaenidae	Papaya, plumeria	Thailand	Saengyot and Burikam (2011)
Ischiodon scutellaris (Fabricius)	Diptera: Syrphidae	Papaya	India	Shylesha et al. (2011a)

Deshpande (2009) recorded the invasion of this pest into cotton-growing tracts of India. The cotton mealybug pest was observed to have high dispersal rate aided by several abiotic factors such as wind, rain and waterways, on farm equipment and

Table 5.5 Parasitoids associated with mealybug, *Paracoccus marginatus*

Species name	Order: family	Host plants	Country	References
Acerophagus papayae (Noyes and Schauff)	Hymenoptera: Encyrtidae	Papaya, plumeria, cassava, cotton	India, Indonesia, Sri Lanka	Shylesha et al. (2011b), Tanwar et al. (2010) and Muniappan et al. (2008)
Anagyrus loecki (Noyes)	Hymenoptera: Encyrtidae	Papaya, plumeria, cassava	India, Indonesia, Sri Lanka, Malaysia	Shylesha et al. (2011b), Tanwar et al. (2010), Nakat et al. (2011) and Muniappan et al. (2008)
Pseudleptomastrix mexicana (Noyes and Schauff)	Hymenoptera: Encyrtidae	Papaya, plumeria, cassava, cotton	India, Indonesia, Sri Lanka, Malaysia, Florida	Shylesha et al. (2011b), Tanwar et al. (2010), Nakat et al. (2011) and Muniappan et al. (2008)
Apoanagyrus californicus Compere	Hymenoptera: Encyrtidae		Mexico, Puerto, Rico	Meyerdirk and Kauffman (2001) and Pantoja et al. (2007)
Pseudaphycus sp.	Hymenoptera: Encyrtidae	Papaya	Mexico	Meyerdirk and Kauffman (2001)
Marietta leopardina	Hymenoptera: Aphelinidae	Papaya	Malaysia	Mastoi et al. (2015)
Chartocerus sp.	Hymenoptera: Signophoridae	Papaya	Malaysia	Mastoi et al. (2015)
Cheiloneurus sp.	Hymenoptera: Encyrtidae	Papaya	Malaysia	Mastoi et al. (2015)

through animals. International trade was believed to have played a major role in the spread of this pest to newer regions of the world.

A number of natural enemies have been recorded on *Ph. solenopsis* (Table 5.6). Parasitoids including *Chalcaspis arizonensis*, *Cheiloneurus* sp., and *Aprostocetus minutus* have been found to parasitize solenopsis mealybug on cotton (*Gossypium* spp.) in Texas, the USA (Fuchs et al. 1991). In India, a solitary endoparasitoid, *Aenasius* sp., was reported to attack *Ph. solenopsis* (Sharma 2007). Hayat (2009) described a new species of parasitoid, *A. bambawalei* (= *arizonensis*), associated with *Ph. solenopsis* with good biocontrol potential. This parasitoid was found parasitizing cotton mealybug, *Ph. solenopsis*, on several plants with the high parasitization efficiency (up to 72%). Parasitism rate of this invasive mealybug on okra (*Abelmoschus esculentus*) was recorded to be 89% (Sharma 2007). *Aenasius arizonensis* was documented to infest 10–45% of the mealybugs on cotton and 5–65% on the alternate hosts. While the parasitism rate of *Paranathrix tachikawai* on cotton mealybugs was found to be 30–39%, *Promuscidea unfasciati* effected as much as 30–80% parasitism rate in nature.

Several species of predators are associated with *Ph. solenopsis*. The predator *Cryptolaemus montrouzieri* has been used several times to control several mealybug pest species. In India, two coccinellids (*Brumoides suturalis* and *Hyperaspis*

Table 5.6 Predators and parasitoids associated with mealybug, *Ph. solenopsis*

Species	Order: Family	Host plants	Country	References
Chalcaspis arizonensis	Encytridae: Hymenoptera	Cotton	Texas, USA	Fuchs et al. (1991)
Cheiloneurus sp.	Encytridae: Hymenoptera	Cotton	Texas, USA	Fuchs et al. (1991)
Aprostocetus minutus	Eulophidae: Hymenoptera	Cotton	Texas, USA	Fuchs et al. (1991)
Aenasius arizonensis	Encytridae: Hymenoptera	Cotton, okra, *Paranathrix tachikawai*	India	Muniappan (2009) and Sharma (2007)
Cryptolaemus montrouzieri, *Brumoides suturalis*, *Hyperaspis maindroni*	Coccinellidae: Coleoptera	Cotton	India	Patel et al. (2009)
Chrysoperla carnea	Chrysopidae: Neuroptera	Cotton	India	Rabinder et al. (2008)

maindroni) were identified to be associated with *Ph. solenopsis* (Patel et al. 2009). Other potential predators, such as the larvae of the lacewing, *C. carnea*, were found to consume 30 mealybug eggs daily in developmental laboratory tests (Rabinder et al. 2008).

5.9.6.3 Natural Enemies in Management of Striped Mealybug, *F. virgata*

Striped mealybug, *F. virgata*, is a polyphagous pest infesting wide variety of crops. Commercially available mealybug destroyer (*C. montrouzieri* Muls.), green lacewings and brown lacewings offer good control over mealybug outbreaks (Franco et al. 2009). Three species of Coccinellidae, two species of Neuroptera and three parasitoid wasp species (*Leptomastix* sp. and two species of *Tetrastichus*) were reported attacking *F. virgata* in Egypt (Awadallah et al. 1979). A coccinellid predator (*Scymnus* sp.) has been reported to attack *F. virgata* in India (Ghose and Paul 1972). Release of *C. montrouzieri* was found effective in controlling the populations of *F. virgata* in guava orchards in India. The local natural enemies, *A. advena* (an encyrtid parasitoid) and *Scymnus coccivora* (a cocinellid predator), also helped in controlling the striped mealybugs (Mani et al. 1990). The caterpillars of Lycaenidae, *Spalgis epius*, were found to be the general predators of several mealybug pests (Chacko and Bhat 1976).

5.9.6.4 Natural Enemies in Management of Pink Mealybug, *M. hirsutus*

The pink hibiscus mealybug, *M. hirsutus*, is a pest of fruit crops and ornamental plants in many parts of Africa, Southeast Asia and northern Australia. A coccinellid, *Chilocorus nigrita* indigenous to the Indian subcontinent and Indo-China has been introduced into many other regions of the world for biological control purpose. Currently, *C. nigrita* is distributed in Bangladesh, Brazil, the Chagos Archipelago, China, Fiji, Ghana, India, Indonesia, Madagascar, Malaysia, Mauritius, Myanmar, Oman, the Pacific, Seychelles, South Africa, Sri Lanka, Thailand, Western Samoa and Yemen (Poorani 2004). The ladybird beetle *C. montrouzieri* is an effective predator on all life stages of *M. hirsutus*. Although *C. montrouzieri* is native to the Australasian zoogeographic region, it has a worldwide distribution due to its introduction in many countries as a potential biological control agent. This beetle is one of the most widely used biological control agents and has been used to target more than 16 pest species (Kairo et al. 2000). *Cryptolaemus montrouzieri* is a polyphagous predator on several groups of bugs belonging to Hemiptera.

5.9.7 Insecticide Resistance and Its Management

In the control of mealybugs, insecticides often play an important role. A relatively large number of insecticides are presently used against mealybugs, including organophosphates, neonicotinoids, carbamates, ketoenols and insect growth regulators. Nevertheless, owing to the elusive nature of mealybug, its characteristic waxy coating on the body and concealed habitats of living, a large number of insecticides are unsuccessful in controlling this group of sucking insect pests. Waxy coatings of mealybug immatures and adults prohibit insecticides from entering them, and insecticide resistance is also developable. Moreover, existing pesticides on the market may not be sufficient to control mealybugs, when they are used only once as it is required to go for a few rounds of pesticide applications. Such repeated use of insecticides allows the insects to develop resistance against the insecticides. As a result, several mealybugs had not been tested by many insecticides. However, multiple insecticide groups contain different active ingredients and can decrease their effectiveness if they develop resistance to the insecticide. Due to the inherent nature of mealybugs, certain insecticides do not properly manage mealybugs.

Inadequate control strategies and exposure of multiple generations of mealybugs accelerate the development of resistance to insecticides. Control failure of insecticides at field level upon the use of the product over a number of years is an indication that the mealybug species has developed resistance to the specific group of insecticide(s).

5.9.7.1 Monitoring of Insecticide Resistance

Although insecticides have been the mainstay of controlling insect pests leading to improvement in agricultural production globally, the use of insecticides has also been restricted by resistance development in several major pests, but some have become pests as a consequence of insecticides being introduced (Mallet 1989). Monitoring of pest populations for their susceptibility status to insecticides is an essential component of resistance management strategies. It would be very helpful to control insecticide resistance in mealybugs to assess the dose reaction of a particular pest population or species to different insecticides. In general, different mechanisms for metabolic resistance operate through a number of detoxification mechanisms elaborated by oxidative and hydrolytic processes to eliminate the toxic effect of insecticides. Increase or decrease in levels of resistance is a dynamic process for insect populations according to selection pressures and exposure regimes (Castle et al. 1996; Horowitz et al. 2002), which eventually alters the presence of resistant populations in the field. Georghiou and Taylor (1986) proposed that economic, microbial, operational and genetic resistances are some of the critical factors determining the development of resistance in pest populations.

Repeated exposure to insecticides favours the individuals to be chosen who are inherently resistant to insecticides to develop resistance for survival. A small portion of the pest population may sustain exposure when a new pesticide is first used due to its distinct genetic makeup. Such individuals pass on this trait to the next generation the genes that are responsible for resistance. Consequent use of the chemical raises the proportion of the population with less resistance. Through such a selection process, resistance to the pesticide steadily develops among the population. Deficiencies in systemic management strategies may also be the reason for resistance growth (Bushra et al. 2014). Various agroecological factors like the prevalence of refuges dilute resistant gene frequencies (Sayyed et al. 2005; Khan et al. 2013). Therefore, knowledge on the vulnerability of the target insect populations to specific insecticides and how they differ between locations can be taken as a guide to assess their genetic potential for the development of resistance. Hence, the spatial variations of susceptibility to insecticides in mealybug populations are to be documented. Development of a diagnostic dose for each test insecticide as deduced from the baseline toxicity data is necessary for regular monitoring of insecticide resistance (Sanderson and Roush 1992; Denholm et al. 1996).

5.9.7.2 Resistance Management and Prevention Strategy

Management strategies to minimize or avoid build-up of resistance enhance the shelf life of existing chemicals for continuous usage (Charles 1996, 2004). The overall strategy is to reduce the selection pressure by optimizing chemical applications, precise insecticidal distribution and rotation of active ingredients of insecticides from different chemical classes against the target group of insects. This is coupled with crop and shelter management strategies aimed at reducing mealybug populations and enhancing insecticide coverage. See below for details:

(i) Identify the mealybug infestations at harvest time. Confine applications to the mealybug habitats. Avoid spraying on crop shelters if the source of pest infestation is properly identified.
(ii) Conserve the available natural enemies in the field.
(iii) Application is to be done based on the economic threshold levels (ETL) of the pest damage. Spray only when pest infestation crosses the ETL.
(iv) If required, follow industry codes of conduct according to label rates.
(v) To get sufficient insecticide coverage, use proper application methods, and follow correct machine speeds and spray conditions. Sprayers should be calibrated at least once per season. Follow instructions for the spray programme beneath the crevices or cracks in trees or fruit bunches or inside the fruit, where they are protected from insecticide contact. High-volume

insecticidal applications are necessary and should be applied to “run-off”; low-volume application technology should not be tried against mealybug pests.

(vi) Identification and understanding of the life cycle of mealybugs are essential. If the most susceptible stage (crawlers) is prevailing, spray insecticides. Use strictly according to label directions to mitigate this threat. Stop exclusive use of same pesticide all season. The capacity for applaud resistance has long been established, and it is usually recommended that it is not used more than twice a year.

(vii) Use different classes of insecticides in rotation if it is required to apply chemicals more than once in a season.

Measures should be taken to assess insecticide resistance across the different mealybug species and geographic populations. Furthermore, sufficient in-depth work should be carried out on biochemical aspects, in particular quantification of molecular resistance mechanisms and detoxifying enzymes by detecting various insecticide resistance alleles, including knock-down resistance, sodium-gated tube and AChe.

5.9.8 Mealybugs as Beneficial Insects

Mealybugs can also be used in biological control of weeds. For controlling *Harrisia* cactus (*Eriocereus martini*), mealybug pest, *Hypogeococcus pungens* Granara de Willink, was tried in Queensland (Australia) as a classical biological control agent through introduction of this insect from Argentina (Williams and Granara de Willink 1992). Many mealybug species have been manipulated in conservation biological control programmes. Cypress trees (*Cupressus* spp.) are widely grown on the borders of crop fields. The cypress mealybug, *Pl. vovae* (Nasonov), serves as an alternative host for natural enemies of mealybug pests of citrus orchards and cocoa plantations (Ho and Khoo 1997; Franco et al. 2004). There are biblical mentions on the use of honeydew excretion of the manna mealybug, *Trabutina mannipara*, as a food source for human beings (Hemprich and Ehrenberg in Ehrenberg) (Ben-Dov 2006; Miller and Kosztarab 1979).

5.10 Conclusion and Future Prospects

A number of morphological, structural and behavioural features render the mealybugs to adapt to diverse ecosystems with a tendency to aggregate and establish themselves in protected sites. Although mealybugs are soft-bodied insects, an impermeable layer of waxy coating makes it hard to penetrate by chemicals, and as such, control of mealybugs by insects is a tough proposition. Anthropological factors and global trade have played a significant part in the recent introductions of several mealybug pests across the continents. Although a number of physical and chemical strategies are being tried against mealybugs, the bioagents have proved to be effective in managing the mealybugs as demonstrated by classical biological control of papaya mealybug, *Pa. marginatus*, or fictitious control of cotton mealybug, *Ph. solenopsis*, or augmentative control of pink mealybug or citrus mealybug by use of a number of parasitoids or predators. Global warming too has contributed to the invasive status of several mealybug pests as these bugs have physiological adaptations to thrive on hot and drought conditions unlike many other sucking insect pests which feed of green succulent plants. Hence, quarantine measures need to be enforced to prevent the spread of invasive mealybugs. Creating awareness on conservative biological control and judicious use of chemicals among farmers would go a long way in effective management of mealybug pests.

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Scale Insects

6

Masumeh Moghaddam, Mousa Abdollahipour, and
Yaghoub Fathipour

Contents

Abstract

Scale insects (Hemiptera: Coccomorpha) attack a huge number of host plants around the world. They generally feed on parenchyma tissue and specially phloem sap. Several factors have caused the scale insects to be considered as important insect pests. They are among the most highly specialized of all plant parasites and feed on all parts of the plant including the roots, stems, leaves, buds, and fruits and may injure or kill plants by feeding on plant sap, injecting toxins, transmitting viruses, or excreting honeydew. Various modes of reproduction (including parthenogenesis and hermaphroditism), rapid population dynamics, frequent lack of host plant specificity (or polyphagy), and typically quite small sizes and concealment allow them to proliferate on various plants in different

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M. Moghaddam
Insect Taxonomy Research Department, Iranian Research Institute of Plant Protection (IRIPP), Agricultural Research Education and Extension Organization (AREEO), Tehran, Iran

M. Abdollahipour · Y. Fathipour (✉)
Department of Entomology, Faculty of Agriculture, Tarbiat Modares University, Tehran, Iran
e-mail: fathi@modares.ac.ir

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habitats and become invasive, especially if control by natural enemies is removed or reduced. Most common scale pests are from three families: the armored scale (Diaspididae), the soft scale (Coccidae), and the mealybug (Pseudococcidae); *Aonidiella aurantii* (Maskell) (Diaspididae), *Planococcus citri* (Risso) (Pseudococcidae), and *Coccus hesperidum* (Linnaeus) (Coccidae) are some of the economically important pests. Integrated pest management (IPM) is a sustainable approach for managing scale insect pests that combines biological, cultural, mechanical, and chemical tools in a way that minimizes economic, health, and environmental risks. The known and available natural enemies for biological control as the safest method for scale insect control are predators especially ladybird beetles (Coccinellidae) and midges (Cecidomyiidae) and wasp parasitoids (mainly Encyrtidae). Other methods such as mating disruption is also used in management programs of scale insects.

Keywords

Coccomorpha · Coccidae · Diaspididae · Pseudococcidae · Monophlebidae

6.1 Introduction

The studying of infraorder Coccomorpha (Williams and Hodgson 2014), which are usually called scale insects, began with Carl Linnaeus' tenth edition of the *Systema Naturae* (Linnaeus 1758). During this period of 260 years, the number of described scale insects has increased from 24 species (Williams 2007) to some 7700 species in more than 1050 genera (García Morales et al. 2016). The root of the word coccidology is derived from the word "*Coccus*," the genus in which Linnaeus included the bulk of his scale insects. Most scale insects were not recognizable as insects by the ancients, but rather as seed or berries and were given the ancient Greek word "Kokkos" and the later Latin word "Coccus" meaning a berry. The word "coccidology," as a branch of entomology, was probably coined for the first time by Tinsley (1899).

Scale insects are sap-sucking hemipterous insects that include all members of the Coccomorpha. These are closely related to aphids (Aphidoidea), whiteflies (Aleyrodoidea), and jumping plant lice (Psylloidea), which make up the suborder Sternorrhyncha (Gullan and Martin 2003). These insects are usually less than 5 mm in length. Their taxonomy is based mainly on the microscopic cuticular features of the adult female. The adult female is paedomorphic, maturing in a juvenile form, whereas the adult male (when present), after going through a prepupal and pupal stage, turns into an alate with nonfunctional mouthparts.

There are currently 54 known scale insect families, of which 35 are extant and 19 are known only as fossils. Scale insects are generally divided into two informal groups, the archaeococcoids and the neococcoids. The archaeococcoids are defined by the presence of 2–8 pairs of abdominal spiracles, such as Margorodidae, Monophlebidae, and Ortheziidae, which are absent in neococcoids. Scale insects are known by various names depending on the family to which they belong, e.g., the armored scales (Diaspididae), mealybugs (Pseudococcidae), putoids (Putoidae), soft scales (Coccidae), ground pearls (Margarodidae), lac insects (Kerriidae), cochineal insects (Dactylopiidae), and ensign scales (Ortheziidae). The most commonly encountered families are those with the most species, namely, Diaspididae, Pseudococcidae, and Coccidae (Gullan and Martin 2003).

Scale insects are small phytophagous arthropods widely distributed in different ecosystems around the world. Their common name derives from the frequent presence of a protective covering (scale) or from the appearance of the insects themselves (Gullan and Cook 2007).

Scale insects are generally phloem sap feeders; however, some feed on parenchyma tissue by directly feeding on the contents of parenchymatic cells. Scale insects are found on various parts of their host and may infest leaves, twigs, branches, and roots, and some live inside plant domatia. Some scale insects are even known to survive on plants completely submerged at high tide (Harrison 1916). Many are important pests of agriculture (Peronti et al. 2001) and may injure or kill plants by depleting them of their sap, injecting toxins, transmitting viruses, or excreting honeydew, which serves as the medium for sooty mold (Williams and Granara de Willink 1992).

Scale insects have been reported as serious pests attacking a huge number of host plants around the world (Miller et al. 2002; Miller 2005). The ongoing and growing transport of timber, live plants, and fruits all around the world is a successful dispersal method for insects with limited mobility. In addition, various modes of reproduction (including parthenogenesis and hermaphroditism), rapid population dynamics, and frequent lack of host plant specificity (or polyphagy) allow many scale insect species to proliferate on various plants in different habitats and become invasive, especially if the control by natural enemies is removed or reduced. The scale insects include many species that attack cultivated plants, and several species, e.g., *Icerya purchasi* Maskell (Monophlebidae), *Maconellicoccus hirsutus* (Green), *Phenacoccus solenopsis* (Tinsley), *Planococcus citri* (Risso) (Pseudococcidae), *Aonidiella aurantii* (Maskell), *Diaspidiotus perniciosus* (Comstock), *Pseudaulacaspis pentagona* (Targioni Tozzetti), and Diaspididae, are economically important as pests of crops (Ouvrard et al. 2013).

Scale insects have been known for centuries, not just for the damage they cause, but for the useful red dyes that some of them produce, for valuable secretions in the form of waxes and resins, and even for their use as medicine and food. Mahdihassan (1954) has given us an account of how lac insects were known to the Chinese in writings dating to AD 320 when the insects produced a red dye and a substance for sticking things together. Lac insects are now known to be tropicopolitan. Scale insects provide other products too. It is generally thought that flower nectar is the main ingredient of honey. However, honeybees collect other sweet ingredients, especially when flowers are scarce. In Greece and Turkey, honeybees collect honeydew from *Marchalina hellenica* (Gennadius) (Marchalinidae) feeding on pine trees, and in Greece alone, this “pine honey” accounts for 60%–65% of all honey produced (Hodgson and Gounari 2006). The honeydew of many species of scale insects in at least six families is known to be a source of bee honey worldwide (Kunkel 1997).

6.2 Host Plants

There are many species of scale insects that feed on a wide range of host plants. Armored scales (Diaspididae) occur on a variety of host plants encompassing more than 1380 plant genera in 182 plant families (Borchsenius 1966).

The most prevalent host families are Leguminosae with about 230 species of armored scale, Poaceae with about 150 species, and Euphorbiaceae with 145 species. Armored scales usually are pests on plants that survive for more than a single year including fruit and nut crops, forest trees, and ornamentals, such as landscape perennials, shrubs, shade trees, and greenhouse plants. Miller and Davidson (1990) compiled a list of 199 species that are considered pests in at least some parts of the world.

Based on an analysis of the host information in the mealybugs catalogue by Ben-Dov (1994), mealybugs occur on species in about 250 families of host plants. The most common host family is Poaceae with 585 species. The Asteraceae is a distant second with 250 species,

As a group, mealybugs attack a very broad range of plants including fruits and vegetables, indoor plant, and outdoor ornamentals like annuals, perennials, shrubs, palms, grasses, and trees.

6.3 Life History

Scale insects are some of the most fascinating and unusual organisms in the Insecta. Their unconventional approach to the plant parasitic regime has produced a myriad of bizarre adaptations. As members of the four-winged Hemiptera, male scale insects are unusual in having the metathoracic wing consistently reduced to stubs (hamulohalteres), and normally, only the mesothoracic wings are well developed. Conversely, as reproductively mature nymphs (neotenic), adult females are wingless and in essence are integumental sacks that function as reproductive factories (Miller and Kosztarab 1979).

All scale insects have similar basic biology, though exceptions occur. Scale insects are sack-like and often do not have functional legs. The term “scale” refers to a substance secreted over the back of the insect. In most cases, adult females are sedentary and do not move. Most species lay eggs underneath their body (ovoviviparous). Some species are parthenogenetic, meaning they can reproduce without matins; others are hermaphroditic; e.g., some species from the genus *Icerya* have both male and female reproductive organs. Females are relatively long-lived, perhaps surviving a number of weeks or months under good conditions. Their main role is to reproduce and they do this very well, often being able to lay hundreds of eggs over their lifetime. Males, when they occur, are very different in appearance to females. They are generally smaller than females and appear superficially similar to whiteflies or gnats. Similar to flies, they only have one pair of functional wings; their hindwings are much reduced. They always have functional legs. Males do not feed, are short-lived, and mainly function to mate with females.

Generally, there are three to four instars in the female and five instars in the male (Miller 2005). First instar nymphs (that either hatch from eggs or are live-born) are called crawlers. Crawlers are the main means through which scale insects naturally disperse. They are generally very small, being about 0.5–1 mm in length, and have functional legs and antennae. They are very lightweight and can disperse with the wind easily. Some species have been reported to be phoretic, meaning the crawler attaches to an insect and "hitch-hikes." After molting to second instar, most scale insects become immobile for the rest of their life; their legs are reduced or absent. They grow in size but are similar in appearance to females. Nymphal females have two or three nymphal instars before becoming an adult. Males always have four nymphal instars, the last of which is metamorphic and called pupa.

All scale insects excrete honeydew, which is basically plant sap and is high in sugar content. Ants may attend scale insects to collect honeydew dropped or shot away from the body of scale insects. Black sooty mold often grows on leaves that have received honeydew.

6.4 Important Pest Scale Insects

Several scale insect families cause economic damage to agricultural crops. The most important pest scale insects are armored scale (Diaspididae), mealybugs (Pseudococcidae), and soft scale (Coccidae).

6.4.1 Family Diaspididae (Armored Scale Insect)

Armored scales are members of the family Diaspididae and are highly specialized plant parasites. The presence of a cover that is not attached to the body makes armored scale easily recognizable in the field. Species occur in all areas of the world with the exception of the polar regions and are common plant pests wherever they are found. Most armored scale pests are invasive in the countries where they cause damage. With increased international trade, these often concealed and cryptic pests pose a serious threat to the world agricultural economy. One-third of the more than 7300 species of scale insects (Coccomorpha) are armored scale (Diaspididae). They are the most species family of scale insects including about 2400 species in 380 genera (Miller and Davidson 2005). Although the diaspidids have been more intensively studied than any other group of coccids, probably not more than half of the existing forms have been recognized and named (Beardsley Jr and González 1975).

Armored scales occur on a variety of host plants encompassing more than 1380 plant genera in 182 plant families (Borchsenius 1966). The most prevalent host

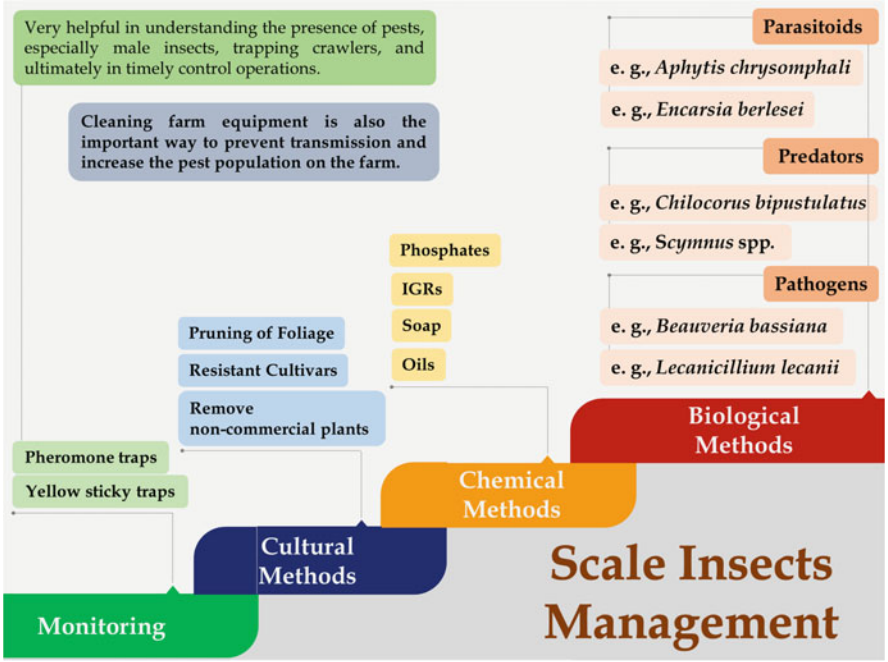

Fig. 6.1 Different control methods of scale insects

families are Leguminosae, Poaceae, and Euphorbiaceae. Armored scales usually are pests on plants that survive for more than a single year including fruit and nut crops, forest trees, and ornamentals, such as landscape perennials, shrubs, shade trees, and greenhouse plants. Miller and Davidson (1990) compiled a list of 199 species that are considered pests in at least some part of the world. This figure is only about 8% of the total number of described species, and their economic impact is quite significant. In the United States, all scale insects are estimated to cause millions (Kosztarab 1977) or even billions (Kosztarab 1990) of dollars in damage and control costs annually. Armored scales are responsible for major portions of these costs (Fig. 6.1).

6.4.1.1 Life History

Armored scales have three female instars and five male instars including adults (Fig. 6.2). Generally, the sexes are indistinguishable morphologically in the first instar. Sexual dimorphism becomes apparent in the second instar. In that stage, differences in the number and distribution of secretory pores, the form of the pygidium, and the development of structures on the pygidial margin are discernible in species in which the sexes have been closely compared (Boratyński 1953). Most are protected by a separate cover that consists of about 50% wax and 50% of a non-waxy component that may be a polyphenol or melaninlike compound (Ebstein and Gerson 1971) and shed skins. Flat strands of wax produced primarily from the

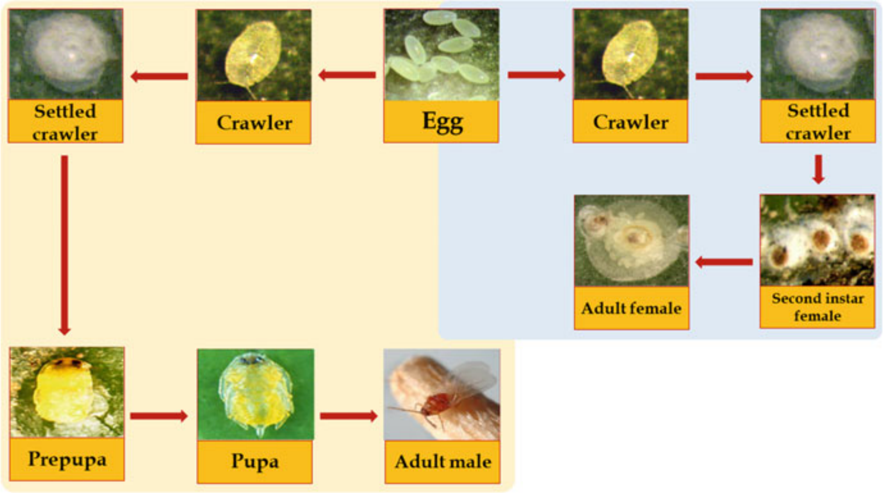

Fig. 6.2 Diaspidid life history chart and scale cover development

pygidial glands form the basic wax component of the cover. It is continuously cemented with material from the anal opening (Foldi 1990).

Most armored scale species are biparental and mating is required for egg production. Other species appear to consist of biparental and uniparental populations, while yet others never seem to produce males (Miller and Davidson 2005). Armored scales live beneath a waxy cover; this cover helps protect them from predators, parasitoids, environmental conditions, and many insecticides. Most species spend the winter in many different life stages including eggs, second instar males and females, or mated adult females. Many species lay eggs under scale cover; a few lay eggs that hatch within minutes of being laid. The first instars, called crawlers, hatch from eggs and are the only mobile stage of armored scales and the only stage without a test. The crawlers' jobs are to find a new spot to feed before settling down to build a test and feed. As you might expect, they can crawl but are also spread around by wind.

The life histories of many of the economically important diaspidids have been worked out in detail. In a particular species, the rate of development and the number of generations per year may vary substantially in different regions. Climatic conditions, particularly temperature, humidity, and rainfall, appear to be the principal controlling factors. California red scale, for example, may have anywhere from two to as many as five or six generations per year (Bodenheimer 1951). Armored scales feed by inserting threadlike hollow mouthparts into leaves or stems and sucking out plant fluids. This removes nutrients and energy that plants need to grow. Thus, scale infestations can reduce plant growth and lead to leaf drop, dead branches, and eventually death.

Among the world's armored scale insects, three high-economic pests broadly distributed across the globe were selected for study.

6.4.1.2 *Aonidiella aurantii* Maskell (Fig. 6.3)

Aspidiotus aurantii Maskell 1879: 199.

Common name: California red scale (CRS).

6.4.1.2.1 Diagnostic Features

Scale cover of adult female is circular, quite flat, translucent yellow-brown, usually stuck to prosoma, and exuviae subcentral. Male scale covers are elongate-oval and paler than that of a female. The first instar forms a white cap.

Slide-mounted adult female shows enlarged prosoma and is heavily sclerotized and strongly reniform at maturity, with prosomal lobes almost enclosing the pygidium. Postsoma is largely membranous and often retracted between prosomal lobes. The pygidium has three pairs of well-developed lobes, the fourth lobes (L_4) each represented by sclerotized prominence. Plates are all fringed. Paraphyses are fusiform, small but distinct, and numbering six pairs. Dorsal macroducts on the pygidium are long and cylindrical and present in different areas: with one between median lobes, two or three submarginal ducts between L_1 and L_2, 5–12 between L_2 and L_3, and 6–18 between L_3 and L_4. Perivulvar pores are absent. The pygidium has two dorsal prevulvar scars and one ventral apophysis on each side.

6.4.1.2.2 Host Plants

Aonidiella aurantii is polyphagous, attacking plant species belonging to more than 177 genera in 83 families (García Morales et al. 2016).

6.4.1.2.3 Distribution

Aonidiella aurantii is widely distributed and has occurred on all zoogeographical regions: Afrotropical, Australian, Nearctic, Oriental, Palaearctic, and subtropical (García Morales et al. 2016). A distribution map of this species was published by CAB International (1996a). This species occurs in all the citrus-growing area of the world, particularly between all latitudes 25 and 40 north and south (DeBach 1962).

6.4.1.2.4 Life History

The California red scale (CRS) is a biparental, ovoviviparous species that may infest all the aboveground parts of host plants (Ferris 1938). The sex pheromone of CRS has been identified and synthesized (Roelofs et al. 1978). Millar and Hare (1993)

Fig. 6.3 *Aonidiella aurantii* (Maskell)

isolated and identified a kairomone from the scale cover, which functions as an oviposition stimulant. CRS males have difficulty finding and mating with CRS females when exposed to an environment with a high concentration of CRS sex pheromone. Crawlers show positive phototropism and tend to travel to the outer canopy, settling on fruits and recent leaves (Campos-Rivela et al. 2012). *Aonidiella aurantii* seems to show a preference for young trees in a good vegetative state, which is where infestations tend to be most severe (Bodenheimer 1951). Almost all males of *A. aurantii* that emerged during a given afternoon were dead the next morning (Mendel et al. 2012).

This scale is a member of a complex species native to the tropics and subtropics of the Old World (Africa through Southeast Asia and the Oriental region) (McKenzie 1937). It became a pest of citrus when introduced into the New World without its associated natural enemies (Compere 1961).

In Algiers climate conditions, the red scale develops three overlapping generations per year on two host plants: (1st) in summer, (2nd) in spring, and (3rd) in autumn. This behavior can be explained by a further study on the influence of trophic factors (Belguendouz-Benkhelfa et al. 2013). The population dynamics showed that there were four male flights including that of the overwintering generation in Spain (Campos-Rivela et al. 2012).

6.4.1.2.5 Nature of Damage

The California red scale is the most important pest of citrus in most citrus-growing areas of the world (Rosen and DeBach 1978). Heavy infestations may cause discoloration, shoot distortion, and leaf drop. The fruit may become pitted and unmarketable. The tree’s bark may split, and the twigs and branches may die back, and this sometimes results in the death of the tree. Chemical control is difficult because the insects are protected by their hard waxy covers. They are also becoming resistant to many insecticides, and indiscriminate use of pesticides has adverse effects on their natural predators.

6.4.1.2.6 Natural Enemies

The aphelinid ectoparasitoids of *Aphytis chrysomphali* (Mercet), *A. lingnanensis*, *A. melinus* DeBach, and *A coheni* DeBach and an aphelinid endoparasitoid of *Encarsia perniciosi* (Tower) as well as the encyrtid endoparasitoid of *Comperiella bifasciata* Howard may control California red scale, if they are not disrupted by pesticides. The coccinellid of *Chilocorus bipustulatus* (L.) is a major predator of this pest (Sorribas and Garcia-Marí 2010). Simultaneous use of both mating disruption and biological control methods can always keep the pest population below the economic injury level (EIL). It also reduces the amount of stains and insect feeding areas on the fruit, which adds to the market value of the product.

6.4.1.3 *Comstockaspis perniciosa* Comstock (Fig. 6.4)

Aspidiotus perniciosus Comstock 1881: 304.

Common name: San Jose scale (SJS).

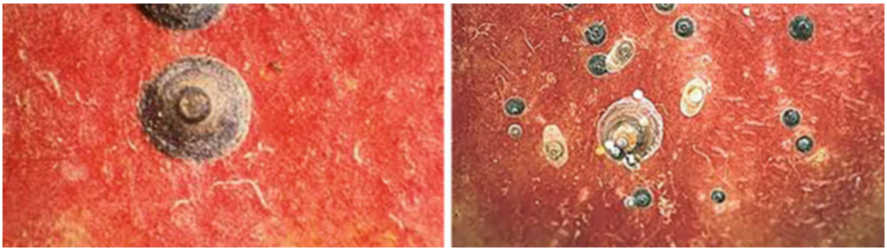

Fig. 6.4 *Comstockaspis perniciosa* (Comstock)

6.4.1.3.1 Diagnostic Features

Female scale cover is light to dark gray, has a diameter of 1.5–2.2 mm, and is more or less circular and slightly convex, with central brown exuviae; the first instar exuviae have a craterlike appearance. Male scale cover is oval with gray exuviae (Kosztarab and Kozar 1988).

The pygidium is round with two pairs of lobes (L_1 and L_2); the third lobes (L_3) are reduced. Bifurcated plates are present between L_1 and L_2 and L_2 and L_3 and laterally to L_3. Dorsal ducts that are 1-barred are present between L_1 and submarginal zones of abdominal segments V–VII; ducts are absent from segments I–IV. Perivulvar pores are absent. Ventral microducts are scattered on the thorax and submarginal zone of abdominal segments.

6.4.1.3.2 Host Plants

Comstockaspis perniciosa is a polyphagous species. The host plant range could probably be much wider. The importance of the host plant is different in different parts of the world, so for each region, only the local literature sources can give a true picture. In general, this species is very dangerous on deciduous fruit trees including apple, pear, peach, plum, currants, and some woody ornamental plants (Kosztarab 1996). Different varieties of fruit species show different susceptibility in different parts of the world, and many fruit species have been studied in this respect (Bichina and Gatina 1976). This scale attacks plant species belonging to more than 71 genera in 42 families (García Morales et al. 2016).

6.4.1.3.3 Distribution

This species is believed to have originated in the area of northern China-Soviet Far East-North Korea (Rosen and DeBach 1978). San Jose scale is widely distributed throughout the temperate and subtropical areas of the world and often is intercepted in quarantine on fruit, especially oranges and tangerines. A distribution map of this species was published by CAB International (1998). *Comstockaspis perniciosa* is widely distributed and has occurred in all zoogeographical regions: Afrotropical, Australian, Nearctic, Neotropical, Oriental, and Palaearctic (García Morales et al. 2016).

6.4.1.3.4 Life History

The San Jose scale is the most studied armored scale insect in the world. According to Rosen and DeBach (1978), this pest may undergo one to five generations per year with the climate being the major limiting factor. In the colder, maritime territories of Russia, one generation per year was reported, but in warmer areas, two (Tereznikova 1969) generations were found. In North America, Johnson and Lyon (1976) reported that up to five generations per year were possible. Occurring ordinarily on the bark, although it may occur on the fruit of apples (Ferris 1938), Iren (1970) studied the symbiosis of this species with the fungi *Septobasidium burtii* and *S. mariani* on apple and pear trees in Turkey. Female sex pheromone has been identified (Gieselmann et al. 1979). This species has three to three and a half generations per year (Wearing 1976), and the females have a high reproductive capacity, each able to produce up to 400 crawlers that disperse and settle on the bark, leaves, and fruit of the trees. Magsig-Castillo et al. (2010) have demonstrated the occurrence of phoretic dispersal of crawlers of *C. perniciosa*. The crawlers use the tarsal and claw digitules of each leg to attach themselves to three different insect species, *Musca domestica* L., *Cryptolaemus montrouzieri* Mulsant, and *Linepithema humile* (Mayr), and can effectively be moved phoretically by these insects.

6.4.1.3.5 Nature of Damage

The San Jose scale is a major pest of deciduous fruit trees in many regions of the world. Its origin is believed to be in the north of the Oriental region, northern China, and Soviet Far East. It was first recorded in United States, in California in 1870, from where it was originally described in 1881. It spread rapidly throughout the main fruit-growing regions of the United States, from west to east. It has since spread to Canada, Central and South America, Europe (from Spain to Caucasus), Japan, India, South Africa, and Australia (Ebeling 1959; Rosen and DeBach 1978; Kozár 1990). This is a pest of kiwifruit in Chile (González 1989). The plant growth regulator heteroauxin was most effective in controlling *C. perniciosa* (Ivanova and Pavlyuchuk 1988). Crop loss caused by *C. perniciosa* on different trees is difficult to assess. The trees lose their vigor and have a shorter life span. Shortly after the introduction to a new country, this pest can infest and kill whole trees and plantations. This species is a quarantine pest in different parts of the world. The import and export of infested fruits or plants could be refused. Local outbreaks have been observed in different parts of the world on fruit trees and ornamental plants, as in Hungary (Kozár and Drozdjak 1988), Switzerland (Kozár et al. 1994), the European part of Russia (Kozár and Konstantinova 1981), Australia (Baker 1977), and Canada (Ker and Sears 1986).

6.4.1.3.6 Natural Enemies

The coccinellid *Chilocorus bipustulatus* as a predator and *Encarsia pernicious* (Tower) and several species of the genus *Aphytis* as parasitoids are the most important biocontrol agents affecting this pest (Bayoumi 2011). Three entomopathogenic fungi are also recommended to control this pest in orchards: *Beauveria bassiana*, *Metarhizium anisopliae* sensu lato, and *Lecanicillium lecanii* with 47% to 77% mortality on San Jose scale (Buhroo 2014). The additional release of different predators and conserving the local natural enemies can increase the efficiency of this type of control.

6.4.1.4 *Pseudaulacaspis pentagona* (Targioni Tozzetti) (Fig. 6.5)

Diaspis pentagona Targioni Tozzetti 1886: 1.

Common name: white peach scale (WPS).

6.4.1.4.1 Diagnostic Features

Female scale is opaque, white, nearly circular, and convex; exuviae has yellow near margin. Male scale is white and faintly tricarinate.

Slide-mounted adult female is broadly turbinate and widest at the mesothorax; prepygidial segments have well-developed lateral lobes. Antennae are set close together. Anterior spiracles are each associated with as many as 18 disc pores. The pygidium has three pairs of lobes; median lobes (L_1) are prominent, and each lobe is notched more than once on each margin and apex rounded; second lobes (L_2) are much smaller than L_1 and pointed or rounded, and inner lobule (L_{2a}) is minute or lacking. Marginal gland spines are present singly on each side of the pygidium and forward to segment V; each gland spine is branched apically, containing two or three microducts. Gland spines are also present in marginal-to-submarginal groups as far forward as abdominal segment 1, and the spines are pointed and becoming progressively shorter. Minute gland spines or duct tubercles are present on the metathorax and prothorax. Dorsal marginal ducts are present on the pygidium; submarginal and submedian segmental rows of macroducts are present on each side of abdominal segment V forward to segment II, progressively decreasing in size. A few minute ducts are also present on ventral margins of the abdominal segment 1 and metathorax. Perivulvar pores are present in five groups.

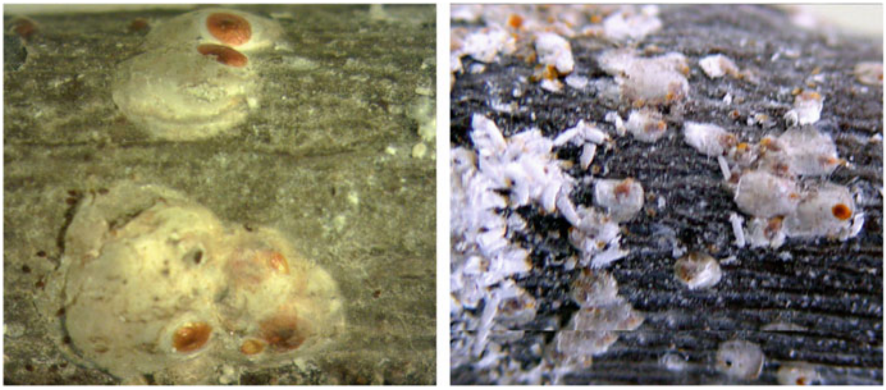

Fig. 6.5 *Pseudaulacaspis pentagona* (Targioni Tozzetti)

6.4.1.4.2 Host Plants

Pseudaulacaspis pentagona is one of the most polyphagous armored scale insect species in the world, except for coniferous plants. Borchsenius (1966) records it from 108 genera in 55 families. In total, *P. pentagona* is found in 221 host plant genera belonging to 85 families (García Morales et al. 2016).

6.4.1.4.3 Distribution

White peach scale is native to eastern Asia and has spread widely to all warmer regions of the world. CAB International (1996b) presented a map of the world distribution of this species. It now occurs in all zoogeographical regions: Afrotropical, Australian, Nearctic, Neotropical, Oriental, and Palaearctic (García Morales et al. 2016).

6.4.1.4.4 Life History

There are two separate generations per year in northern Switzerland. The flight of the males of the first generation starts in the middle/at the end of June and continues until the middle/end of July; the flight of the second generation lasts from the middle/end of August to the end of September/middle of October. Due to adverse climatic conditions, the flight of the second generation often is poor (Mani et al. 1997). Each female lays between 100 and 150 eggs, depending largely on host plant species. Male eggs are orange, and female eggs are white. The eggs hatch three or four days after being laid, and the first instars actively swarm over the host, searching for a suitable feeding site. The females have three instars, and males have five instars. There are one generation to four generations per year, depending upon climate, although in the United Kingdom only one generation is most likely. In the United States, a generation is completed in 36 to 40 days during the summer at an average temperature of 25 °C and in 80 to 90 days during the winter. The adult females overwinter and can survive temperatures as low as −20 °C although there is high mortality at such temperatures. Sheble and Kozár (1995) found two generations in Hungary. The beginning of the first generation (usually in the first week of June) did not show considerable variation from that in Italy and southern France (Kosztarab and Kozar 1988).

6.4.1.4.5 Nature of Damage

Beardsley Jr and González (1975) list white peach scale as one of the 43 principal armored scale pests of the world. Johnson and Lyon (1976) note that this scale was very destructive to ornamental trees and shrubs. This scale as a common pest of peach has become the most important pest of peach in Iran (Bazrafshan et al. 2010). WPS is a pest of economic importance for mulberry, trees, and woody ornamental. In the United States, crop losses from WPS plus control costs were only $94,000, but on the untreated peach trees, the yield loss was valued at $480,000 (Kosztarab 1990). This species was a serious pest of mulberry trees and a menace to the silk industry and neighboring countries (Rosen 1990).

6.4.1.4.6 Natural Enemies

There are many biocontrol agents used to control white peach scale. An aphelinid ectoparasitoid, *Aphytis* sp., and an aphelinid endoparasitoid, *Encarsia berlesei* (Howard), as well as two efficient predators, *Chilocorus bipustulatus* (L.) (Coccinellidae) and *Cybocephalus fodori minor* (Endrody-Younga) (Cybocephalidae), are the most useful biocontrol agents of *P. pentagona* worldwide (Saber et al. 2010).

6.4.2 Family Pseudococcidae

The family Pseudococcidae, commonly known as the mealybugs, is the second largest family within the scale insects (Hemiptera: Sternorrhyncha: Coccomorpha), with 2001 described species in 260 genera (García Morales et al. 2016). Mealybugs are named for their powdery waxy secretions covering the bodies of the immature stages and adult wingless females. Although this covering is usually white, it is somewhat variable and provides a few diagnostic characters in life. Many mealybug species form an ovisac in which they lay the eggs but some bear live young ones. Some species are parthenogenetic, but in sexually reproducing species, the adult males are normally minute, without functional mouthparts, and are generally winged but are occasionally apterous. The current taxonomy and classification of the mealybugs are based on the morphology of adult female, although a few studies have included adult males.

Mealybugs are mostly phloem-sucking plant parasites and can be found on almost any part of a plant. A few are virus vectors, and many species are extremely important economic pests, attacking agricultural and horticultural crops, forestry, and ornamental plants. Some of them are very important agricultural pests in the world, for example, the citrus mealybug *Planococcus citri* (Risso), the pink hibiscus mealybug *Maconellicoccus hirsutus* (Green), and the solenopsis mealybug *Phenacoccus solenopsis* Tinsley.

6.4.2.1 Life History

In mealybugs, the female has three nymphal instars, whereas the male has four instars (first, second, third or prepupal, and fourth or pupal). Mealybugs are slow moving and feed on plant sap. Mealybugs tend to live in aggregations, often on the underside of leaves, on stems, around petioles and branches, and other concealed areas, such as in dense foliage, where fruits are touching or leaves overlap and near flower buds and growing tips. Mealybugs feeding on grasses and grasslike plants are often present under the leaf sheath. Some species may be on aboveground parts and on roots; other species are solely root feeding. Unlike most other scale insects, subsequent nymphal stages and adults have functional legs and are generally able to move short distances. Nymphs are similar to females but smaller and will molt through three to four nymphal instars. However, male nymphs can be more elongated than female nymphs and have four to five nymphal stages. Males also have a pupal stage before emerging as adults. Generally, mealybugs complete their life cycle in 1–3 months, depending upon temperature and host plant. Therefore, 4–10 generations can occur each year.

6.4.2.2 *Planococcus citri* (Risso) (Fig. 6.6)

Dorthesia citri Risso 1813: 416.

Common name: citrus mealybug.

6.4.2.2.1 Diagnostic Features

The oval, flat body of the female is wingless, distinctly segmented, and about 3 mm (0.12 inch) in length. Waxy secretions make it look as if it is covered with flour; they also form a fringe of long filaments. The winged males appear about 4.5 mm (0.17 inch) long due to their tail filaments.

The body of an adult female is oval. Antennae are eight segmented. Legs are well developed. Translucent pores are present on hind coxae and tibiae. Cerarii numbering is 18 pairs, each bearing two conical setae, except for preocular pairs each sometimes with 1–3 conical setae. Circulus is normally quadrate and divided by an intersegmental line. Ostioles are well developed. Dorsal and ventral setae are flagellated and ventral ones are longer. Cisanal setae are shorter than anal ring setae. Multilocular disc pores are present on venter only, in rows across abdominal segment III and posterior segments. Trilocular pores are evenly distributed on dorsum and venter. Discoidal pores are sparse on both dorsum and venter. Dorsal oral collar ducts don't have apparent rims and are larger than ventral ducts, often present next to some cerarii; ventral oral collar ducts are of two sizes, small ducts across median areas of abdominal segments and larger ducts around margins including the head and thorax, and are scattered in median area of the thorax.

6.4.2.2.2 Host Plants

Planococcus citri attacks a wide range of crop plants, ornamental plants, and wild flora. In the literature, 195 genera belonging to 83 host plant families are named for this mealybug (García Morales et al. 2016).

Fig. 6.6 *Planococcus citri* (Risso)

6.4.2.2.3 Distribution

The pest is a native of Asia but is also found throughout the Americas, Europe, and Oceania (CABI/EPPO 1999). At present, it is found in 159 countries in all zoogeographical regions (García Morales et al. 2016).

6.4.2.2.4 Life History

Eggs are deposited in white, cottony masses, called ovisacs, on the trunk and stems of citrus plants, giving the appearance of cotton spread on the plant. The glossy, light yellow eggs are oval and approximately 0.3 mm long. A female can lay from 300 to 600 eggs in her lifetime, and eggs are deposited in groups of 5 to 20. Depending on the season, egg hatch may occur after six to 10 days or several weeks. Female lays an average of 29 eggs per day (Meyers 1932). Nymphs emerge from the ovisacs and typically settle along midribs and veins on the underside of leaves, young twigs, and fruits. They can also be found where two fruits are touching each other or on leaves clinging to fruits. Due to their habit of hiding in crevices, light infestations are easily overlooked. Wax and honeydew secreted by crawlers are visible indicators of infestations. First instar female and male nymphs are called crawlers. The nymphs take six to ten weeks to reach maturity. The nymphs are yellow, oval-shaped with red eyes, and covered with white, waxy particles (Griffiths and Thompson 1957). The female nymphs resemble the adult females, while male nymphs are more elongated. Female nymphs have four instars. Males differ greatly; they have three instars and a prepupal stage. It is only the males that can produce a cottony-appearing cocoon and pupate (Anonymous 2007). Female mealybugs are wingless and, therefore, must be transported to subsequent host plants, although they are able to crawl for short distances. The immatures can be blown by the wind. Females can live for up to 29 days depending on the host plant. Mealybugs remain motile throughout their life cycle, except for the male pupa (Anonymous 2007; Griffiths and Thompson 1957). This mealybug is most common during the spring and early summer. Several overlapping generations occur in a year, but only one cycle (spring–summer) is of major concern to citrus growers in Florida, with peak infestations occurring in June or July. With the advent of the rainy season and warm weather, populations rapidly decline. In grapefruit groves, mealybugs persist in high numbers throughout the summer and into the fall. Reproduction in the greenhouse can occur year-round, leading to continuous populations of mealybugs (Griffiths and Thompson 1957).

6.4.2.2.5 Nature of Damage

This is one of the most cosmopolitan mealybugs, damaging many outdoor crops in the tropics and subtropics, as well as in greenhouse in the temperate regions (Cox 1989). This is a sporadic olive pest in the Mediterranean region (Swirski 1985). Rosciglione and Castellano (1985) showed that this mealybug transmitted grapevine virus A (GVA) from grapevine to *Nicotiana clevelandii*. Lagowska (1995) reviewed the prospects for biological control in greenhouses. Hanna et al. (1956) reported that *P. citri* is a vector of the swollen-shoot virus disease of cacao in Ghana. Cabaleiro and Segura (1997) studied in Spain the field transmission of grapevine leafroll-associated virus 3 (GLRaV-3) by the mealybug *P. citri*. It is very likely that the studied mealybug was *P. ficus* (Signoret).

6.4.2.2.6 Natural Enemies

The use of natural enemies cannot greatly reduce pest damage, especially at the beginning of the season and when young fruits are attacked, although there have been successful application of them against the pest. The encyrtid *Anagyrus pseudococci* as a parasitoid; the brown lacewing (*Sympherobius sanctus* Tjeder) (Hemerobiidae), larvae of *Dicrodiplosis* spp. (Cecidomyiidae), *Nephus* spp., *Scymnus* spp., and *Cryptolaemus* spp. (Coccinellidae) as predators; and some entomopathogenic nematodes are the important control agents of *P. citri*. Insect growth regulators (IGRs) and biocontrol agents are not recommended to be used at the same time, as they can cause high mortality in predators (Michelakis and Hamid 1995).

6.4.2.3 *Maconellicoccus hirsutus* (Green) (Fig. 6.7)

Phenacoccus hirsutus Green 1908: 25.

Common name: pink hibiscus mealybug.

6.4.2.3.1 Diagnostic Features

Adult mealybugs are small (about 3 mm long) and pink in body colour but covered with a waxy secretion. The waxy filaments are short and females are usually obscured by this white mealy wax. When adults are crushed, their body fluids are also pink coloured. Adult males are smaller than females, reddish brown and have one pair of wings. Males have two long waxy "tails."

The mounted adult female is broadly oval. Anal lobes are poorly to moderately developed, with each ventral surface bearing an anal lobe bar. Antennae are nine segmented each. Legs are well developed, and claw doesn't have a denticle. Translucent pores are present on posterior surface of the hind femur and tibia. Cerarii are usually numbering 4–6 pairs. Anal lobe cerarii has two short conical setae each and a few trilocular pores, all situated on a membranous area. Anterior cerarii is each similar to anal lobe cerarii but sometimes reduced to a single seta or with one or both

Fig. 6.7 *Maconellicoccus hirsutus* (Green)

setae replaced by flagellate setae. Circulus are present, quadrate to oval, and divided by an intersegmental line. Ostioles are well developed. Dorsal and ventral setae are flagellated; ventral setae are usually longer than those on dorsum. Multilocular disc pores are present on venter, mainly across anterior and posterior edges of abdominal segments as far forward as abdominal segment IV, often reaching margins, sometimes present on abdominal segment III and the head. Trilocular pores are evenly distributed on dorsum and venter. Oral rim ducts are numerous, numerous across dorsal segments and margins of the thorax and abdominal segments, and ventrally on marginal areas on the head, thorax, and abdomen. Oral collar ducts are of two sizes, both narrower than trilocular pores: small pores across the middle of dorsal segments in single rows, sometimes reduced to only one or two per segment, and on venter across the middle of abdominal segments to margins and on the head and thorax and large pores present ventrally across abdominal segments III–VI and on lateral margins of all abdominal segments, plus a few in medial and marginal areas of the thorax.

6.4.2.3.2 Host Plants

Maconellicoccus hirsutus attacks a wide range of predominantly woody plants, including many ornamentals. This mealybug occurs on 222 genera belonging to 78 host plant families (García Morales et al. 2016).

6.4.2.3.3 Distributions

Maconellicoccus hirsutus is native to southern Asia and has spread to other parts of the world. From its center of origin, *M. hirsutus* has spread to the rest of Asia, Africa, the Caribbean, Hawaii, and the Americas (Kairo et al. 2000). Currently, *M. hirsutus* is distributed in 93 countries and territories, and its distribution continues to expand in South America. To date, no established infestation has been detected in Europe (EPPO 2005).

6.4.2.3.4 Life History

The life cycle of *M. hirsutus* has been studied in India. Each adult female lays 15–600 eggs over a period of about one week, and these hatch in 6–9 days (Bartlett 1978b). A generation is completed in about five weeks in warm condition. In countries with a cool winter, the species survives cold conditions as eggs (Bartlett 1978b) or other stages, both on the host plant and in the soil (Pollard 1995). There may be as many as 15 generations per year (Pollard 1995). Small “crawlers” are readily transported by water, wind, or animal agents. Crawlers settle in cracks and crevices, usually on new growth which becomes severely stunted and distorted, in which densely packed colonies develop. There are three immature instars in the female and four in the male. Reproduction is mostly parthenogenetic in Egypt (Hall 1921), but *M. hirsutus* is biparental in West Bengal (Ghose 1971, 1972) and probably in the Caribbean (Williams 1996). Infestations of *M. hirsutus* are often associated with attendant ants (Mani 1989).

6.4.2.3.5 Nature of Damage

Similar to many phloem-feeding insects, *M. hirsutus* produces a copious amount of honeydew, which reduces the aesthetic value of plants and provides a growing medium for black sooty mold, which further reduces value and normal growth and reproduction. *Maconellicoccus hirsutus* releases a plant growth regulator-type toxin during feeding, which results in severe stunting, decline, and deformation of growing terminals, leading to “bunchy top” symptoms in sensitive plants and premature senescence of flowers and foliage. Accidental introductions of *M. hirsutus* have caused significant economic damage to several Caribbean countries. In the late 1990s, estimated economic losses (including direct crop losses, cost of control operations, and impact on trade) were US$18.3 million in Grenada, US$5.1 million in Trinidad, and US$3.4 million in the Grenadines (Kairo et al. 2000). Although Guyana did not suffer direct crop losses, the loss of trade in regional export markets was considerable. By comparison, infestation in Puerto Rico was under successful biological control, which led to reduced crop injury and avoided significant economic losses (Michaud 2003). The potential economic impact of *M. hirsutus* to US agriculture was estimated at US$750 million annually and US$5 billion over 10 years if left uncontrolled, with greatest impacts in the ornamental and vegetable markets (Moffitt 1999). A more recent estimate projected the annual damage to avocado (*Persea americana*), citrus (*Citrus* spp.), cotton (*Gossypium hirsutum*), peanut (*Arachis hypogaea*), soybean (*Glycine max*), nursery, and vegetable crops at US$163 million in Florida or US$1.6 billion for the entire United States (Ranjan 2006). Outside of North America, *M. hirsutus* has emerged as a serious pest of cotton, *Gossypium* spp., in India and Pakistan (Muralidharan and Badaya 2000), possibly due to fewer insecticide applications on crops engineered to express *Bacillus thuringiensis* toxins for caterpillars.

6.4.2.3.6 Natural Enemies

The encyrtid parasitoids *Anagyrus kamali* and *Gyranusoidea indica* Shafee, some predators including Neuroptera and Cecidomyiidae, and various Coccinellidae species such as *Cryptolaemus montrouzieri* and *Scymnus* spp. are active agents on pink hibiscus mealybug worldwide (Roltsch et al. 2006).

6.4.2.4 *Phenacoccus solenopsis* Tinsley (Fig. 6.8)

Phenacoccus solenopsis Tinsley 1898: 47.

Common name: cotton mealybug (CMB).

6.4.2.4.1 Diagnostic Features

The adult female usually has patches of dark grey cuticle bare of white mealy wax, with three pairs of submarginal patches on the abdomen and one pair on the thorax.

The body of adult female is broadly oval. Anal lobes are well developed. Antennae each has nine segments. Legs are well developed; claw has a denticle. Translucent pores are present on the hind tibia. Cerarii are numbering 18 pairs. Anal lobe cerarii each are with two lanceolate setae and a few trilocular pores; anterior cerarii are similar. Circulus is usually oval, sclerotized, and flaccid. Ostioles are

Fig. 6.8 *Phenacoccus solenopsis* Tinsley

normal, with inner edges of the lips, and only lightly sclerotized. Dorsal multilocular disc pores are absent; ventral multilocular disc pores are present posterior to vulva and usually in a single row medially along posterior edges of abdominal segments VI–VII and anterior edge of segment VII. Oral collar ducts are not numerous in medial areas across abdominal segments V–VII, anterior to multilocular disc pores, and also with one or two tubular ducts usually present near margins of some posterior abdominal segments and posterior to each anterior spiracle.

6.4.2.4.2 Host Plants

The solenopsis mealybug has been recorded on 201 host plant species belong to 63 families that include field crops, ornamentals, trees, and vegetables (García Morales et al. 2016).

6.4.2.4.3 Distributions

The solenopsis mealybug has a wide geographical distribution with its origin in Central America (Williams and Granara de Willink 1992) followed by reports from the Caribbean and Ecuador (Ben-Dov 1994), Chile (Larrain 2002), Argentina (Granara de Willink 2003), Brazil (Gulik and Gullan 2005), and Iran (Moghaddam and Bagheri 2010). The distribution records include all zoogeographic regions of the world (García Morales et al. 2016).

6.4.2.4.4 Life History

Females of *P. solenopsis* are ovoviviparous, bisexual species that have been reported as capable of producing from 150 to 600 eggs, protected within a waxy ovisac (Lu et al. 2008). Upon hatching, females undergo three immature stages prior to reaching adulthood, whereas males undergo first, second, prepupa, and pupa stages prior to adulthood. The period of development from crawler to adult stage is approximately 25–30 days, depending upon the weather and temperature. This species is capable of producing multiple generations annually. The biology study of CMB in India showed that parthenogenesis with ovoviviparity (96.5%) was dominant over the oviparous (3.5%) mode of reproduction. Males accounted for less than 5% of the population (Vennila et al. 2010). Cotton mealybug can complete 15 generations per year in the field as reported by Tanwar et al. (2007). The field biological study of cotton mealybug on Chinese hibiscus shrubs in Ahvaz, Iran, is completed in 11 generations per year (Seyfollahi et al. 2017). The population consists of overlapping generations, and individuals of various developmental stages can be observed on the same time (Hameed et al. 2012).

6.4.2.4.5 Nature of Damage

The solenopsis mealybug is an important plant pest worldwide (Williams and Granara de Willink 1992; Hodgson et al. 2008). Mealybug feeding may cause the leaves to turn yellow and results in defoliation, reduced plant growth, or plant death. The presence of the solenopsis mealybug has the potential to inflict significant damage to field crops, i.e., cotton (*Gossypium* spp.) and tobacco (*Nicotiana* spp.), in all growing periods of the plants. This mealybug caused serious damage to cotton in Pakistan in 2005 (Saeed et al. 2007; Dhawan et al. 2009) and India (Jhala et al. 2008; Bhosle et al. 2009). Also, it is a pest of commercial crops including a variety of vegetables, grapes (*Vitis vinifera*), jute (*Corchorus* spp.), mesta (*Hibiscus cannabinus*), and tobacco.

The economy of Pakistan is heavily dependent on the production of cotton. Cotton production is reported to account for 8.2% of the value added in agriculture and 2% of the GDP of Pakistan. The significant damage to cotton caused by *P. solenopsis* has a significant impact on the economy of the nation. Economic crop losses of an estimated 14% occurred in Pakistan in 2005 and Punjab, India, in 2005–2006 and 2006–2007 (Hodgson et al. 2008). In the 2005 growing season, this invasive pest was responsible for a 44% reduction in seed cotton yields in Pakistan (Dhawan et al. 2009). The intense attack by the mealybug on Bt cotton resulted in significant economic losses to growers in the Punjab region (Dutt 2007). In 2007, the number of hectares committed to growing cotton increased over the previous year, but cotton production had a significant decline over the previous year (Dutt 2007). Previous field crop losses in cotton have ranged from 30% to 80% in some regions of India (Nalwar et al. 2009). However, the grower is required to implement chemical applications to save the crop, resulting in increased expenses in production as well as the potential of chemical contamination of soil and water.

6.4.2.4.6 Natural Enemies

The most important control factor for this pest is an encyrtid endoparasitoid, namely, *Aenasius arizonensis*, which can control up to two-thirds of the pest population. Tactics suggested to suppress populations of the invasive mealybugs on crops include allowing the parasitoid to build up in population numbers prior to applying chemical insecticides (Pala Ram and Saini 2010). The use of beneficial predators provides the opportunity to control *P. solenopsis* on cotton (*Gossypium* spp.) in several countries. The predator, *Cryptolaemus montrouzieri*, has been imported to India and released in cotton-infested fields as a control tool for *P. solenopsis*. The coccinellid predators *Brumoides suturalis* (Fabricius) and *Nephus regularis* Sicard are other natural enemies of this pest (Spodek et al. 2018). To provide a higher probability of success by implementing natural control agents, several species of ants that are often associated with honeydew-producing mealybugs and protect their population from natural enemies would need to be controlled (Tanwar et al. 2007).

6.4.3 Family Coccidae

The family Coccidae, commonly known as the soft scale insect, is the third largest family within the scale insects (Hemiptera: Sternorrhyncha: Coccomorpha), with 1217 described species in 174 genera (García Morales et al. 2016). Adult female soft scales are usually 3–9 mm in length and often occur in groups that are easily seen and collected. Soft scales have several distinctive features, which aid in their recognition in the field. With the possible exception of very old or very convex individuals, most soft scales have two pairs of white, button-like wax structures on the body margin. One pair will be found about the middle of each side; another pair will be found anterior to these. These wax buttons are at the outer end of a furrow which connects each ventral thoracic spiracle with the body margin. The furrows allow air to reach the spiracles since the rest of the venter of the scale is closely pressed against the host surface. Adult soft scales usually grow considerably in size and often change drastically in appearance between maturation and the time that oviposition is complete and the scale dies. Most young adult females are oval in shape, being narrow anteriorly and broader posteriorly, but some species are circular, elliptical, elongate, or deltoid. In most species, the young adults are flat in profile and press the venter as close as possible to the host surface until oviposition begins. Some soft scales produce white waxen covers or ovisacs over the eggs and do not protect the eggs under the body.

The family is recognized by a pair of triangular anal plates (opercula) and by the anal cleft which splits the body from the anal plates to the posterior body margin. The anal plates occur in all stages except the egg, pupal, and adult male stages and in the adult females of *Physokermes*. Most species have two pairs of differentiated spiracular setae associated with the thoracic spiracles. The spiracular setae are located at the outer ends of the spiracular furrows along the body margin. The spiracular furrows are usually delimited by a band of quinquelocular pores.

Many soft scales cause economic injury to ornamental plants and fruit and nut crops. Although a few species are known to kill their hosts, many causes reduced host vigor, productivity, defoliation, and dieback. Most are phloem feeders and produce large amounts of honeydew. The honeydew is a growth substrate for sooty mold fungi which give the plants an unsightly appearance and is thought to interfere with the photosynthetic processes to hosts (Gill 1988).

6.4.3.1 General Life History

The majority of soft scales appear to be parthenogenetic although males occasionally develop in some species. A few species, such as *Eulecanium tiliae*, are obligatorily biparental. Females of most species undergo five stages including the eggs, first instar nymph (crawler), second nymphal instar, third nymphal instar, and adult. Most males undergo six life stages including the eggs, first instar nymph (crawler), second nymphal instar, prepupal stage, pupal stage, and adult. Most soft scales have one annual generation. Eggs and crawlers are produced in the spring and summer. Most species probably overwinter as third stage nymphs and reach the adult stage in early spring. A few species such as brown soft scale, *Coccus hesperidum*, have multiple generations. Most soft scales have well-developed legs in all stages and are capable of moving from one location to another. Many soft scales that prefer to feed on the leaves of deciduous hosts during the summer months must move to the twigs and branches before winter leaf drop (Gill 1988).

6.4.3.2 *Ceroplastes floridensis* Comstock (Fig. 6.9)

Ceroplastes floridensis Comstock 1881: 331.

Common name: Florida wax scale.

6.4.3.2.1 Diagnosis

Adult female Florida wax scales are elliptical and reddish brown with a short anal process (Hamon and Williams 1984) and range from 2 to 4 mm in length and 1 to 3.5 mm in width. Each female has a reddish body that is coated with a thick layer of pinkish-white wax. Males are not known in this species (Futch et al. 2009).

Slide-mounted adult female is broadly oval. Stigmatic clefts are shallow. Caudal process is conical, pointing dorso-posteriorly. Length is about 1.0–3.5 mm and width 0.8–2.5 mm. Dorsal derm is membranous except for heavily sclerotized caudal process. Derm has seven clear areas, and medio-dorsal area is absent. Dorsal setae are very short. Preopercular pores are in a narrow band. Marginal setae are all curved, each stoutly setose with a sharp apex; abundant stigmatic clefts are shallow, each with a line of rather bullet-shaped stigmatic setae extending some distance along margin on either side of cleft. Ventral derm is entirely membranous. Pregenital disc pores are abundant around genital opening and across the preceding segment. Spiracular disc pores are in a fairly narrow band of 50–90 pores. Ventral tubular ducts, with a small tufted glandular apex, are present in a distinct band extending from near each antenna to near each anal lobe. Antennae each have six (rarely seven) segments. The legs are well developed, each without a tibiotarsal articulatory sclerosis and each claw without a denticle; claw digitules are both broad.

Fig. 6.9 *Ceroplastes floridensis* Comstock

6.4.3.2.2 Host Plants

The Florida wax scale has been recorded on 152 genera belonging to 67 host plant families (García Morales et al. 2016).

6.4.3.2.3 Distribution

Ceroplastes floridensis has an almost worldwide distribution in tropical, subtropical, and Mediterranean areas, having been recorded from the Austro-Oriental, Neotropical, New Zealand and South Pacific, Nearctic, and Oriental regions, as well as the Afrotropical region (Hodgson and Peronti 2012).

6.4.3.2.4 Life History

Three generations of Florida wax scale occur in Florida (Johnson and Lyon 1991), but two generations per year are common throughout its global range. Each generation lasts about three to four months. The first generation occurs in April and May, the second is in July and August, and the third one occurs in October and November. There are three instars (Drees et al. 2006). First instars (crawlers) hatch after two to three weeks of egg development, emerge from underneath the female, and disperse and settle on other leaves, stems, and twigs to begin feeding and secreting wax around their bodies. Those scales that settle on the leaves often align themselves along the leaf midrib (Drees et al. 2006). Older nymphs can move around within the same plant to search for new flushes of growth on which to feed. Florida wax scales can also overwinter as newly mature females (Drees et al. 2006) and develop two annual generations on *Citrus* in Greece (Argyriou and Kourmadas 1980). Two generations per year developed in Queensland, Australia (Smith et al. 1997).

6.4.3.2.5 Nature of Damage

The direct damage is caused by nymphs inserting their mouthparts into the plant tissue and withdrawing large amounts of plant fluids. Heavy infestations can discolor leaves, cause premature leaf drop, and branch dieback. Plant death is also possible. As a result of the scales consuming so much plant fluid, they excrete a considerable amount of a sugary, sticky honeydew, which then becomes colonized by a sooty mold fungus (Argov et al. 1987).

The sooty mold can cause a significant reduction in photosynthesis and aesthetic value (Hodges et al. 2000). Other insects are also attracted to and feed on the honeydew, including various bees, paper wasps, hornets, velvet ants, imported fire ants, and other ant species.

6.4.3.2.6 Natural Enemies

Hymenopterous endoparasitoids *Aprostocetus ceroplastae*, *Coccophagus lycimnia* (Walker) (Aphelinidae), *Metaphycus eruptor* Howard (Encyrtidae), and *Scutellista cyanea* Motschulsky (Pteromalidae), the coccinellid *Chilocorus bipustulatus*, and spiders are the most important biocontrol agents that attack young scales of this pest species (Tavares et al. 2019). Potassium nitrate and IGRs, which do not harm the natural enemies, and organophosphates, carbamates, and systemic insecticides cause high death on the pest population.

6.4.4 Family Monophlebidae

The family Monophlebidae is commonly known as the giant scales (Hemiptera: Sternorrhyncha: Coccomorpha), with 262 described species in 47 genera (García Morales et al. 2016). Adult female body is large, up to 10 mm or more, and generally elongate-oval; the legs and antennae are usually conspicuous and dark. They occur on the stems, branches, or foliage of the plant, usually with wax covering the body, occasionally without wax, and often form an ovisac or a marsupium.

6.4.4.1 General Life History

Giant scales have relatively simple life history patterns compared with cyst-forming margarodoids. In general, they have four female instars and five male instars, but unlike most other scale insects, the prepupa is quite mobile, and although it may have wing buds, the legs and antennae are well developed (Morales 1991). *Drosicha mangiferae* Green has one generation each year. Eggs hatch in December or January after diapausing in ovisacs in the soil or the duff around the host. First instars move to the leaves and molt three times to become adults. Males are indistinguishable from females until the third instar prepupa. The prepupa wanders for a while, forms a waxy test, and molts to the pupal stage. Adults appear in April, mate, and migrate off of the host to the ground where an ovisac is produced and eggs are laid. The life cycle of *Icerya purchasi* Maskell is similar, but developing females lay their eggs directly on the host in a fluted ovisac that is attached to the body of the adult female. Males are uncommon. Females are hermaphrodites that frequently inseminate themselves. Adult males mate with females, but it is not clear if their sperms are used for reproduction (Hughes-Schrader 1930). There are two or three generations each year depending on the climate.

6.4.4.2 *Icerya purchasi* Maskell (Fig. 6.10)

Icerya purchasi Maskell 1879: 221.

Common name: Australian fluted scale; cottony cushion scale.

6.4.4.2.1 Diagnosis

The adult female scale has a reddish-brown body, black legs, and antennae. Tufts of short, black hairs occur in parallel rows along the edge of the body. However, the

Fig. 6.10 *Icerya purchasi* (Maskell)

most distinguishing characteristic of this scale is the large, elongated, grooved cottony-white egg sac. The egg sac (10 to 15 mm in length) becomes 2 to 2.5 times as long as the body of the female. Each egg sac will contain 600 to 1000 bright-red eggs.

Adult female is oval to pyriform in shape, 7–9 mm long, and yellowish orange in color, and its legs, antennae, and eyes are dark brown. Antennae are 11 segmented, and apical segment is the longest. The eyes are simple and conical. Tufts of short, black hairs occur in parallel rows along the edge of the body. Thoracic and abdominal spiracles has a bar, lacking disc pores at opening and in the atrium. Small pores with rims of four or five loculi and open centers are present on the head, thorax, and abdomen. Band of ovisac pores with rims of 6–9 loculi and 1 inner loculus extends across the first abdominal segment and continues around body margin. Fine, black setae have two sizes. Ventral cicatrices are posterior to the vulva, numbering three; the median one is the largest. Cicatrices has a granular or reticulate surface texture. Long, fine, black setae and derm hairs are numerous and grouped on middorsal areas of the head and thorax; fine and stout short derm hairs are present and anal tube as for subfamily (Morales 1991).

6.4.4.2.2 Host Plants

Icerya purchasi has been recorded on host plants belonging to 167 genera and 68 families (García Morales et al. 2016).

6.4.4.2.3 Distribution

Cottony cushion scale is widely distributed and occurs in 127 countries from all zoogeographical regions: Afrotropical, Australian, Nearctic, Oriental, Palaearctic, and subtropical (García Morales et al. 2016).

6.4.4.2.4 Nature of Damage

Cottony cushion scale extracts plant sap from leaves, twigs, and branches, reducing tree vigor. If infestations are heavy, leaf and fruit drop can occur along with twig dieback. It also secretes honeydew, which promotes the growth of sooty mold that may discolor fruit and block photosynthesis. *Icerya purchasi* became one of the most economically important insect pests of citrus soon after its discovery in the United States on *Acacia* at Menlo Park, California, in 1868 or 1869 (Ebeling 1951). Since then, *I. purchasi* has been recorded as a pest of citrus and ornamentals in over 80 countries and appears to be most abundant in tropical and semitropical regions of the world (Hale 1970).

6.4.4.2.5 Natural Enemies

The control of the cottony cushion scale is always referred to as the first successful example of biological control of pests by an introduced natural enemy. The cryptochaetid endoparasitoid *Cryptochaetum iceryae* Williston can be used as adjuncts to *Rodolia cardinalis* for biological control. The positive performance of this parasitoid has been seen in various countries. The parasitoid *Cryptochaetum iceryae* has also proved to be effective in regulating *I. purchasi* populations. Adults of *C. iceryae* are sensitive to heat and aridity and are most effective in regulating cottony cushion scale populations in cooler coastal areas. In more arid and hot areas, vedalia beetles are more effective. Studies in California (Quezada and DeBach 1973) have shown that the two natural enemies seasonally share their prey in different proportions and are fairly even in their competitive abilities, *Rodolia* usually taking more prey during summer and autumn and *Cryptochaetum* taking more during winter and early spring. Competition between the two natural enemies did not increase host survival, and these studies provide strong support for the importation of multiple natural enemies. Other control agents have been applied against the pest, but they have not been successful (Hoddle et al. 2013).

6.5 Nature of Damage

Scale insects have been reported as serious pests attacking a huge number of host plants around the world (Miller et al. 2002; Miller 2005; Kondo et al. 2008). These insects are found on various parts of their hosts and may infest leaves, twigs, branches, and roots, and some live inside plant domatia (Kondo et al. 2008). They feed almost exclusively on the phloem of their host plants to which they cause direct damage, but they can also cause indirect damage by transmitting plant pathogens through infection or through the buildup of honeydew, promoting the attack of plant pathogens (Ross et al. 2010). Kosztarab (1990) reviewed the effects of scale insects feeding on plants and recognized 11 types of damage depending on the scale species involved and its preferred feeding site. Damage symptoms generally are most evident when feeding occurs on leaves or fruits. Species such as *Unaspis euonymi* on euonymus and *Aonidiella aurantii* on citrus remove chlorophyll in a circle around the scale and create a yellow halo. The species *Pseudaulacaspis cockerelli* on the palm feed in one direction on a leaf forming a narrow chlorotic area (called a stylet track). This track may become darker with age. Many scale species prefer to feed on the lower surface of leaves, but their presence is normally evidenced by the chlorotic blotches that are visible on the upper leaf surfaces. A few leaf-feeding species produce reddish halos, such as *Diaspidiotus perniciosus* (San Jose scale), on several hosts. Regardless of the scale species or the feeding symptoms, heavily infested leaves usually turn yellow or brown and die and fall from the host.

There have probably been outbreaks of scale insects causing damage to local crops and plants for centuries, but the arrival in the United States of *Icerya purchasi* Maskell (Monophlebidae) toward the end of the nineteenth century, resulting in the almost collapse of the citrus industry, seemed to attract attention throughout the world. Outbreaks causing considerable damage are occurring to the present day, and in the last 40 years, the accidental introduction of the cassava mealybug *Phenacoccus manihoti* Matile-Ferrero (Pseudococcidae) from South America to West Africa caused considerable damage to cassava throughout Africa affecting the staple food of 200 million people (Herren and Neuenschwander 1991). In recent times, the hibiscus mealybug *Maconellicoccus hirsutus* (Green) was introduced accidentally to the Caribbean area affecting a large number of plant species including fruit trees and plants of economic importance (Chang and Miller 1996). The pest species were brought under control by parasitoids or predators with the aid of taxonomists who could identify the pest species accurately and suggest areas where natural enemies could be located. More recently, the lobate lac scale *Paratachardina pseudolobata* Kondo and Gullan has caused serious damage in Florida, the Bahamas and Christmas Island, Australia (Kondo and Gullan 2007; Schroer et al. 2008).

6.6 Management Measures

6.6.1 Integrated Pest Management

The management of pest scale insects requires a combination of tactics including insecticide application and biological control (Fig. 6.1 and Table 6.1). Successful examples often involve specific predators or parasitoids, but the fight against polyphagous invasive species may be impeded by a lack of knowledge about their systematics. Accurate identification of a pest provides a set of knowledge about its biology, host preference, preferred sites of feeding, area of origin, and natural enemies (Ouvrard et al. 2013).

The several stages for successful management of scale insects are as follows.

6.6.1.1 Monitoring

Monitoring is essential for all pests, diseases, and general plant health. Monitoring provides essential information for making management decisions. Monitoring for scale insects should include visual observations including checking for the presence of egg masses, crawlers, and parasitized individual. This is important as it indicates that populations will increase quickly if management actions are not taken. The proportion of different life stages present, most notably crawlers, may also change the exact actions taken; crawlers are susceptible to pesticides, but other stages are more tolerant.

6.6.1.2 Cultural Management

(i) Inspect all plant parts, including roots, for any signs of pests; (ii) monitor plants regularly; (iii) only propagate from clean mother stock plants; (iv) remove plants with heavy infestation; (v) remove crop debris and disinfest the growing area after removing a consignment of plants that have had scale insects with a suitable product; (vi) provide an optimal growing environment, including appropriate nutrition, water, growing media, and other conditions; (vii) control ants as they spread crawlers and protect scale insects from natural enemies; (viii) keep the growing area and surroundings free of weeds; and (ix) ensure adequate plant spacing. This allows greater air movement and increases pesticide coverage.

Table 6.1 The list of important natural enemies of scale insects with their associated families.

Natural enemy			
Family	Species	Scale insect family	References
Predators			
Coccinellidae	*Hyperaspis notata*	Pseudococcidae	González and Gordon (2009)
Cecidomyiidae	*Diadiplosis* sp.	Pseudococcidae	Harris (1968)
Coccinellidae	*Rodolia cardinalis*	Monophlebidae	Bartlett (1978a)
Coccinellidae	*Cryptolaemus montrouzieri*	Coccidae, Monophlebidae, Pseudococcidae	Panis (1981)
Parasitoids			
Aphelinidae	*Aphytis lepidosaphes*	Diaspididae	DeBach and Landi (1961)
Aphelinidae	*Coccophagus lycimnia*	Coccidae	Lampson and Morse (1992)
Aphelinidae	*Aphytis chilensis*	Diaspididae	Alexandrakis and Neuenschwander (1980)
Encyrtidae	*Mariola flava*	Aclerdidae, Pseudococcidae	Noyes (2010)
Encyrtidae	*Anagyrus saccharicola*	Pseudococcidae	Noyes (2017)
Encyrtidae	*Mucrencyrtus aclerdae*	Aclerdidae	De Santis (1972)
Encyrtidae	*Pauridia peregrina*	Coccidae	Salazar (1972)
Encyrtidae	*Metaphycus lounsburyi*	Coccidae	Blumberg and Swirski (1982)
Encyrtidae	*Metaphycus helvolus*	Coccidae	Blumberg and Swirski (1982)
Encyrtidae	*Metaphycus flavus*	Coccidae	Abd-Rabou (2001)
Encyrtidae	*Diversinervus elegans*	Coccidae	Viggiani and Mazzone (1977)

6.6.1.3 Chemical Control

Scale insects are difficult to manage using pesticides alone. Contact products tend to slough off their body, and they may be tolerant to systemic products. If pesticides are to be used to manage scale insects, it is recommended to apply contact products only when there is a high proportion of crawlers present. Crawlers are very susceptible to many pesticides, including oil-based products. When high populations are present, a systemic product will probably be required.

6.6.1.4 Biological Control

There are a number of natural enemies that can manage scale insect including commercially available predatory insects and naturally occurring parasitoid wasps and predators. There are also a range of fungi and bacteria that may infect and kill scale insects, although these are less likely to substantially reduce populations unless they become very abundant.

6.7 Conclusions

Scale insects can cause significant damage to agriculture and especially to gardening due to their high reproductive ability and specific morphology. Despite their small size, if these pests are not controlled in time, they can even completely destroy the plant. Therefore, using different control methods such as biological, chemical, and mechanical measures can help the farmers control their populations. This chapter has attempted to provide useful information on important species of scale insects along with their biological characteristics and methods of control and management.

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Gram Pod Borer (*Helicoverpa armigera*)

7

Geetanjali Mishra and Omkar

Contents

Abstract

Helicoverpa armigera, a polyphagous pest, has traditionally been prevalent throughout the old world. While it has been predicted that it has the potential to make its foray into the New World, till very recently, it was not a reality. However, with its advent into southern America in recent years, the chances of its entry into northern America are extremely high. In such a scenario, with its wide host range of 181 species belonging to 47 families, many of which are major agricultural crops, the likelihood of this pest causing severe global damage is very high. *Helicoverpa armigera* is well on its way to becoming a ubiquitous pest. This chapter discusses the biology, damage measures and management strategies for this pest, which is currently causing a crop loss of USD 78 billion globally annually.

G. Mishra (✉) · Omkar
Department of Zoology, University of Lucknow, Lucknow, India

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7.1 Introduction

The genus *Helicoverpa* (Lepidoptera: Noctuidae) includes some of the most harmful and wide-ranging polyphagous lepidopteran pests of agriculture globally (Cunningham and Zalucki 2014; Kriticos et al. 2015; Sosa-Gomez et al. 2016). The extant species are *Helicoverpa armigera* (Hübner), *Helicoverpa assaulta* (Guenée), *Helicoverpa atacamae* Hardwick, *Helicoverpa fletcheri* Hardwick, *Helicoverpa gelotopoeon* (Dyar), *Helicoverpa hardwicki* Matthews, *Helicoverpa hawaiiensis* Quaintance and Brues, *Helicoverpa helenae* Hardwick, *Helicoverpa pallida* Hardwick, *Helicoverpa prepodes* Common, *Helicoverpa punctigera* Wallengren, *Helicoverpa toddi* Hardwick, *Helicoverpa virescens* (Fabricius) and *Helicoverpa zea* Boddie. Of these, *H. armigera*, *H. zea* and *H. punctigera* are responsible for the maximum global crop damage. Each of these species is highly polyphagous and largely attacks high-value agricultural and horticultural crops. Of the three species, *H. armigera* is most widespread across the globe with historical ranges of Africa, Asia, Europe and Australasia and the Americas in recent years added to the new range (Jones et al. 2019; Table 7.1).

The highly polyphagous nature of this noctuid moth can be deciphered from the reported 181 plant species that are spread across 47 families, on which it feeds (Manjunath et al. 1989; Zalucki et al. 1994; Karimi et al. 2012). Many of these plant species form some of the food staples globally, such as chickpea, soybean, maize, groundnut, cowpea and tomato. Other than known plant species, its wide food palate also makes it likely for *H. armigera* to continue adding more plants to its already vast food range.

It can cause 21% to 36% crop yield losses in Ethiopia (Geletu et al. 1996) and almost 30% in India (Dinesh et al. 2017) to 37–50% in Pakistan (Khan and Faizullah 1999). Chaudhary and Sharma (1982) reported that a single larva of *H. armigera* damaged 7–10% pods causing 5.4% yield loss and damaged 6.7% pods and 6.2% grains per meter row length. Yield loss of 400 kg/ha by pod borer was observed during favourable weather conditions, and damage could reach up to 90–95% (Rahman 1990; Sachan and Katti 1994). It has been estimated that the global losses by *H. armigera* alone are approximately USD 78 billion (Kriticos et al. 2015).

What makes this noctuid moth a highly potent pest and great global concern are its polyphagous food range and three unique life history features of high mobility via facultative migration, high fecundity and facultative diapause. These life history traits make it possible for this moth to survive in volatile ever-changing habitats and adapt promptly to seasonal fluctuations, thus making it a 'global pest'. The ability to spread via long-range migration (Farrow and Daly 1987; Feng et al. 2004, 2005a, b, 2009; Gregg et al. 1995; Pedgley 1986; Pedgley et al. 1987; Raulston et al. 1982, 1986) and international trade (Tay et al. 2017) has together turned this moth into a global threat of massive proportions. This potent mix of distinctive life history features results in the ability of this particular moth to develop insecticide resistance at an alarming rate (Joussen et al. 2012; Xia et al. 2014), which further adds to the vortex of problems that this species creates towards our global agricultural and horticultural yield. In view of the enormous agricultural losses that this moth causes,

Table 7.1 Global distribution of *Helicoverpa*

Continent/country	Distribution	Reference
Africa		
Algeria	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Angola	Present	UK, CAB International (1993) and EPPO (2014)
Benin	Present	UK, CAB International (1993) and EPPO (2014)
Botswana	Present	UK, CAB International (1993) and EPPO (2014)
Burkina Faso	Present	UK, CAB International (1993) and EPPO (2014)
Burundi	Present	UK, CAB International (1993) and EPPO (2014)
Cabo Verde	Present	UK, CAB International (1993) and EPPO (2014)
Cameroon	Present	UK, CAB International (1993) and EPPO (2014)
Central African Republic	Present	UK, CAB International (1993) and EPPO (2014)
Chad	Present	UK, CAB International (1993) and EPPO (2014)
Democratic Republic of the Congo	Present	UK, CAB International (1993) and EPPO (2014)
Republic of the Congo	Present	UK, CAB International (1993) and EPPO (2014)
Côte d'Ivoire	Present	UK, CAB International (1993) and EPPO (2014)
Egypt	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Eritrea	Present	UK, CAB International (1993)
Eswatini	Present	UK, CAB International (1993) and EPPO (2014)
Ethiopia	Present	UK, CAB International (1993) and EPPO (2014)
Gabon	Present	UK, CAB International (1993) and EPPO (2014)
Gambia	Present	UK, CAB International (1993) and EPPO (2014)
Ghana	Present	UK, CAB International (1993) and EPPO (2014)
Guinea	Present	UK, CAB International (1993) and EPPO (2014)
Kenya	Present	UK, CAB International (1993) and EPPO (2014)
Lesotho	Present	UK, CAB International (1993) and EPPO (2014)
Libya	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Madagascar	Present	UK, CAB International (1993) and EPPO (2014)
Malawi	Present	UK, CAB International (1993) and EPPO (2014)
Mali	Present	UK, CAB International (1993) and EPPO (2014)
Mauritania	Present	UK, CAB International (1993) and EPPO (2014)
Mauritius	Present	UK, CAB International (1993) and EPPO (2014)
Mayotte	Present	EPPO (2014)
Morocco	Present, localized	UK, CAB International (1993) and EPPO (2014)
Mozambique	Present	UK, CAB International (1993) and EPPO (2014)
Namibia	Present	UK, CAB International (1993) and EPPO (2014)
Niger	Present	UK, CAB International (1993) and EPPO (2014)
Nigeria	Present	UK, CAB International (1993) and EPPO (2014)
Réunion	Present	UK, CAB International (1993) and EPPO (2014)

(continued)

Table 7.1 (continued)

Continent/country	Distribution	Reference
Rwanda	Present	UK, CAB International (1993) and EPPO (2014)
Saint Helena	Present	UK, CAB International (1993) and EPPO (2014)
Senegal	Present	UK, CAB International (1993) and EPPO (2014)
Seychelles	Present	UK, CAB International (1993) and EPPO (2014)
Sierra Leone	Present	UK, CAB International (1993) and EPPO (2014)
Somalia	Present	UK, CAB International (1993) and EPPO (2014)
South Africa	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Sudan	Present	UK, CAB International (1993) and EPPO (2014)
Tanzania	Present	UK, CAB International (1993) and EPPO (2014)
Togo	Present	UK, CAB International (1993) and EPPO (2014)
Tunisia	Present, localized	UK, CAB International (1993) and EPPO (2014)
Uganda	Present	UK, CAB International (1993) and EPPO (2014)
Zambia	Present	UK, CAB International (1993) and EPPO (2014)
Zimbabwe	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Asia		
Afghanistan	Present	UK, CAB International (1993) and EPPO (2014)
Armenia	Present	UK, CAB International (1993) and EPPO (2014)
Azerbaijan	Present	UK, CAB International (1993) and EPPO (2014)
Bangladesh	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Bhutan	Present	UK, CAB International (1993) and EPPO (2014)
Brunei	Present	Waterhouse (1993)
Cambodia	Present	UK, CAB International (1993), Waterhouse (1993) and EPPO (2014)
China	Present, localized	UK, CAB International (1993) and EPPO (2014)
Cocos Islands	Present	EPPO (2014)
Georgia	Present	UK, CAB International (1993) and EPPO (2014)
Hong Kong	Present, widespread	EPPO (2014)
India	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Indonesia	Present	UK, CAB International (1993) and EPPO (2014)
Iran	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Iraq	Present	UK, CAB International (1993) and EPPO (2014)
Israel	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Japan	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Jordan	Present	UK, CAB International (1993) and EPPO (2014)

(continued)

Table 7.1 (continued)

Continent/country	Distribution	Reference
Kazakhstan	Present	UK, CAB International (1993) and EPPO (2014)
Kuwait	Present	UK, CAB International (1993) and EPPO (2014)
Kyrgyzstan	Present	UK, CAB International (1993) and EPPO (2014)
Laos	Present, widespread	UK, CAB International (1993), Waterhouse (1993) and EPPO (2014)
Lebanon	Present	UK, CAB International (1993) and EPPO (2014)
Malaysia	Present	UK, CAB International (1993) and EPPO (2014)
Myanmar	Present	UK, CAB International (1993) and Waterhouse (1993); EPPO (2014)
Nepal	Present	UK, CAB International (1993) and EPPO (2014)
North Korea	Present	UK, CAB International (1993) and EPPO (2014)
Pakistan	Present	APPPC (1987), UK, CAB International (1993) and EPPO (2014)
Philippines	Present	UK, CAB International (1993) and Waterhouse (1993) and EPPO (2014)
Saudi Arabia	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Singapore	Present	UK, CAB International (1993), Waterhouse (1993) and EPPO (2014)
South Korea	Present	UK, CAB International (1993) and EPPO (2014)
Sri Lanka	Present	UK, CAB International (1993) and EPPO (2014)
Syria	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Taiwan	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Tajikistan	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Thailand	Present	UK, CAB International (1993), Waterhouse (1993) and EPPO (2014)
Turkey	Present, localized	UK, CAB International (1993); EPPO (2014)
Turkmenistan	Present	UK, CAB International (1993) and EPPO (2014)
United Arab Emirates	Present	UK, CAB International (1993) and EPPO (2014)
Uzbekistan	Present	UK, CAB International (1993) and EPPO (2014)
Vietnam	Present	UK, CAB International (1993), Waterhouse (1993) and EPPO (2014)
Yemen	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Europe		
Albania	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Austria	Present, few occurrences	EPPO (2014)

(continued)

Table 7.1 (continued)

Continent/country	Distribution	Reference
Belgium	Absent, intercepted only	EPPO (2014)
Bulgaria	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Croatia	Absent, formerly present	EPPO (2014)
Cyprus	Present, localized	UK, CAB International (1993) and EPPO (2014)
Czechia	Absent, eradicated	EPPO (2014) and CABI (Undated-a or Undated-b)
Denmark	Absent, intercepted only	UK, CAB International (1993) and EPPO (2014)
Estonia	Absent, formerly present	EPPO (2014)
Finland	Present, few occurrences	EPPO (2014)
France	Present, localized	UK, CAB International (1993) and EPPO (2014)
Germany	Present, few occurrences	UK, CAB International (1993) and EPPO (2014)
Greece	Present, widespread	UK, CAB International (1993), EPPO (2014) and CABI (Undated-a or Undated-b)
Hungary	Present, localized	UK, CAB International (1993), Bozsik (2007) and EPPO (2014)
Italy	Present, localized	UK, CAB International (1993) and EPPO (2014)
Latvia	Absent, formerly present	EPPO (2014)
Lithuania	Present	Ostrauskas et al. (2002)
Malta	Present, localized	UK, CAB International (1993) and EPPO (2014)
Moldova	Present	Timus and Croitoru (2006)
Montenegro	Present	Radonjić and Hrnčić (2011)
Netherlands	Absent, eradicated	IPPC (2007) and EPPO (2014)
North Macedonia	Present	EPPO (2014)
Norway	Absent, formerly present	UK, CAB International (1993) and EPPO (2014)
Poland	Present, few occurrences	UK, CAB International (1993) and EPPO (2014)

(continued)

Table 7.1 (continued)

Continent/country	Distribution	Reference
Portugal	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Romania	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Russia	Present, localized	EPPO (2014)
Serbia	Present, localized	EPPO (2014)
Serbia and Montenegro	Present, localized	UK, CAB International (1993)
Slovakia	Present, localized	EPPO (2014)
Slovenia	Present, widespread	EPPO (2014)
Spain	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Sweden	Present	Palmqvist (2015)
Switzerland	Present, localized	UK, CAB International (1993) and EPPO (2014)
Ukraine	Present, widespread	UK, CAB International (1993) and EPPO (2014)
United Kingdom	Absent, eradicated	EPPO (2014)
North America		
Puerto Rico	Present, localized	NAPPO (2014)
USA	Absent, formerly present	NAPPO (2016) and NAPPO (2015)
Oceania		
American Samoa	Present	UK, CAB International (1993) and EPPO (2014)
Australia	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Federated States of Micronesia	Present	EPPO (2014)
Fiji	Present	UK, CAB International (1993) and EPPO (2014)
Guam	Present	EPPO (2014)
Kiribati	Present	UK, CAB International (1993) and EPPO (2014)
Marshall Islands	Present	UK, CAB International (1993) and EPPO (2014)
New Caledonia	Present, widespread	UK, CAB International (1993) and EPPO (2014)
New Zealand	Present, widespread	UK, CAB International (1993) and EPPO (2014)
Norfolk Island	Present	Holloway (1977), UK, CAB International (1993) and EPPO (2014)

(continued)

Table 7.1 (continued)

Continent/country	Distribution	Reference
Northern Mariana Islands	Present	UK, CAB International (1993) and EPPO (2014)
Palau	Present	EPPO (2014)
Papua New Guinea	Present	APPPC (1987), UK, CAB International (1993) and EPPO (2014)
Samoa	Present	UK, CAB International (1993) and EPPO (2014)
Solomon Islands	Present	UK, CAB International (1993) and EPPO (2014)
Tonga	Present	UK, CAB International (1993) and EPPO (2014)
Tuvalu	Present	UK, CAB International (1993) and EPPO (2014)
Vanuatu	Present	UK, CAB International (1993) and EPPO (2014)
South America		
Argentina	Present, localized	EPPO (2014) and Murúa et al. (2014)
Brazil	Present, localized	EPPO (2014)
Paraguay	Present, localized	EPPO (2014) and Murúa et al. (2014)
Uruguay	Present	Castiglioni et al. (2016)

Sourced from CABI 2020, https://www.cabi.org/isc/datasheet/26757

there is immense need for developing an understanding about this moth, the reasons for its spread and latest measures that can be employed for the management of its population. In this chapter, we will discuss the identifying features, spread, nature of damage and the control measures employed for the control of *H. armigera.*

7.2 Distribution

Helicoverpa armigera is an old-world moth. It is found in the Palearctic, Oriental, Ethiopian and Australian regions, in tropical, dry and warm temperate climates (CAB 2000; Jones et al. 2019; Fig. 7.1). Wubneh (2016) mentions that this noctuid moth is most abundant in deserts and xeric shrublands, Mediterranean scrub, temperate broadleaf and mixed forests, tropical and subtropical grasslands, savannas and shrublands, and tropical and subtropical moist broadleaf forest.

Till the early 2000s, this moth was unheard of in either South or North America, where traditionally *H. zea* has had a foothold. Though *H. armigera* had been recognized as a serious threat in North America with models predicting a 49% potential of establishment, it has till now been largely kept at bay due to strict screening at ports of entry of produce from across the globe. Many models have indicated that northern Brazil could connect to North America via Caribbean islands (Kriticos et al. 2015, Zalucki and Furlong 2005). Studies have indicated a regular interception of *H. armigera* bearing cut flowers and herbs from old-world countries (Venette et al. 2003; Pogue 2004). It is anticipated that compared to *H. zea*,

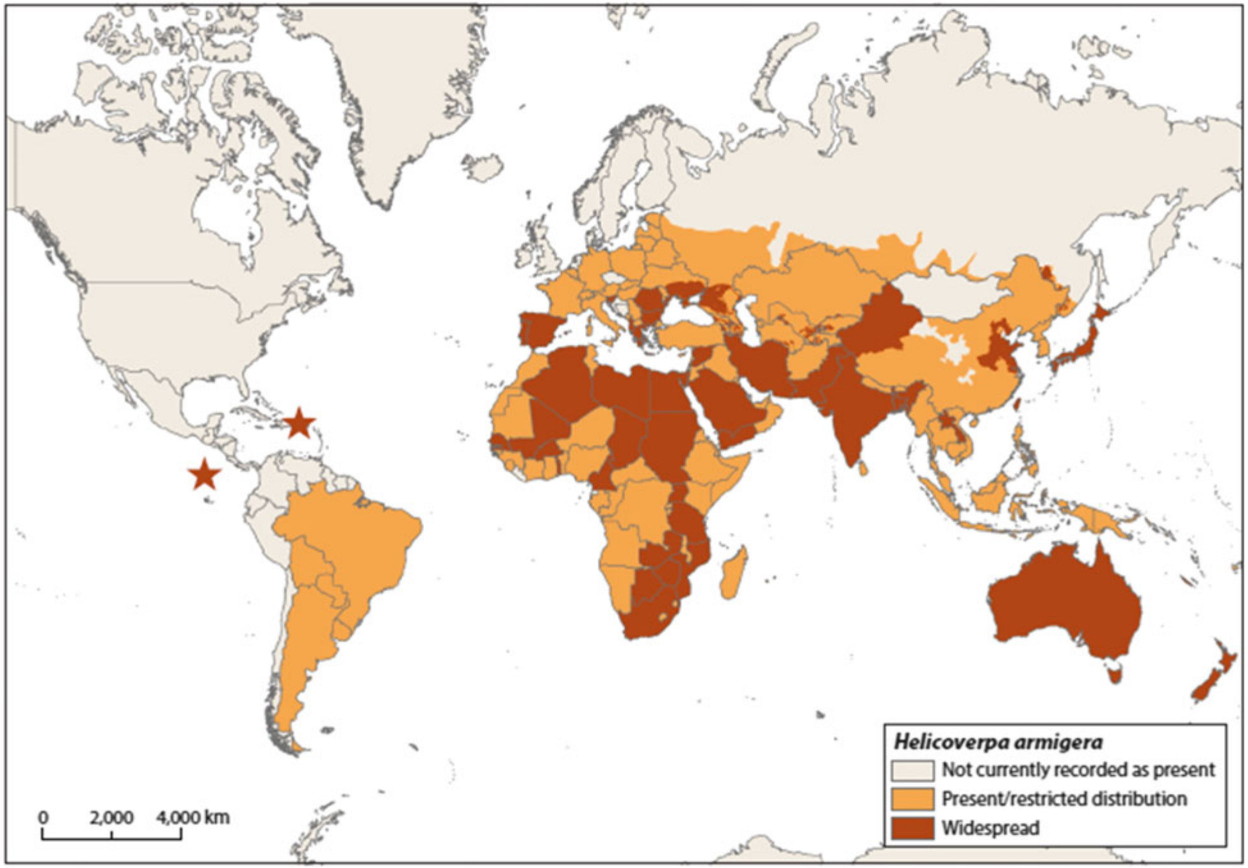

Fig. 7.1 Distributionof *H. armigera* across the globe. (Jones et al. 2019)

H. armigera is more likely to cause massive damage to the corn and cotton crops (Venette et al. 2003; Pogue 2004). While North America is still defended against the scourge of this pest, in Brazil, it is a quarantine pest since 1999 and A1 quarantine pest since 2008 (Oliveira et al. 2003; Lima et al. 2006). It has since its introduction into the New World (Tay et al. 2013) expanded its range in Central and South America.

Too-cold regions are not likely to assist the establishment of *H. armigera* populations, since their pupa does not overwinter very successfully. However, global warming might reverse this current trend and lead to further expansion of their range. Studies across various species of *Helicoverpa* have indicated that *H. armigera* has high genetic diversity which probably is the major contributing factor behind its high adaptability and invasiveness (Anderson et al. 2018).

7.3 Host Plants

Helicoverpa armigera has a wide range of host plants, which include many crop plants, such as cotton, sorghum, sunflower, chickpeas, lucerne, lupins, soybeans, tobacco, maize and wheat, and horticultural crops, such as tomatoes, lettuce, capsicum, various bean crops and flowers: chrysanthemums, gladioli and roses (Kriticos et al. 2015). Thirty-five plant families have been reported as hosts of *H. Armigera* in Australia (Zalucki et al. 1986, 1994). In India, extensive studies have been conducted on host plant range, with 181 species recorded from 47 families (Manjunath et al. 1989). Global records include 68 families, but only 14 families are commonly consumed across the globe (Cunningham and Zalucki 2014).

While the immature stages feed on various plant parts, the survival and development of larvae are best on flowers and fruiting bodies (Liu et al. 2010; Soleimannejad et al. 2010), which is in turn responsible for the massive economic loss. It is owing to the consumption of fruiting bodies that most common names of *Helicoverpa* include the terms budworm, bollworm, earworm, etc.

7.4 Life History

Helicoverpa armigera is a holometabolous insect, which means that it has four major stages, the egg, larva, pupa and adult. To suitably manage pest populations, it is essential to conclusively identify all life stages and also to understand the life history and modulation of the pest. We will first discuss the identifying features of each stage and then the life cycle and its modulations due to different abiotic and biotic factors.

7.4.1 Egg

The eggs are yellowish white and gleaming when freshly laid. With the passage of time, as the larva develops inside the egg and the yolk is consumed, these eggs gradually turn dark brown just prior to hatching. The eggs are pomegranate shaped and 0.4–0.6 mm in diameter. The apical area has micropyles and is smooth, unlike the rest of the egg surface that is ribbed with approximately 24 longitudinal ribs and multiple bisecting transverse ribs. This gives the appearance of a corn cob (Fig. 7.2a).

7.4.2 Larva

There are six larval instars in *H. armigera*, which means that they undergo five moults. (http://download.ceris.purdue.edu/file/3616). The first instars when freshly emerged are translucent and yellowish white and darken in colouration with age. At emergence, while the rest of the body is yellowish white, the head, prothoracic shield, supra-anal shield, prothoracic legs, spiracles and raised base of the setae are dark brown to black, giving rise to an apparently spotted appearance (King 1994; Bhatt and Patel 2001). Second instars are yellowish green in colour and have black thoracic legs. Five abdominal prolegs are present on the third to sixth and tenth abdominal segments in both first and second instars.

The full-grown larvae are long, ventrally flattened and dorsally convex. The final body segment is elongated. Full-grown larvae are approximately 30–40 mm in length. They possess a brown mottled head, pale brown prothoracic and supra-anal plates, black claw and spiracles and minute closely placed tubercles. They are highly variable in colouration ranging from brownish, to reddish, to pale green (Yamasaki et al. 2009). There are characteristic marking patterns: a narrow, dark, median dorsal band followed by a pale broad band and a dark broad band on each side. The lateral

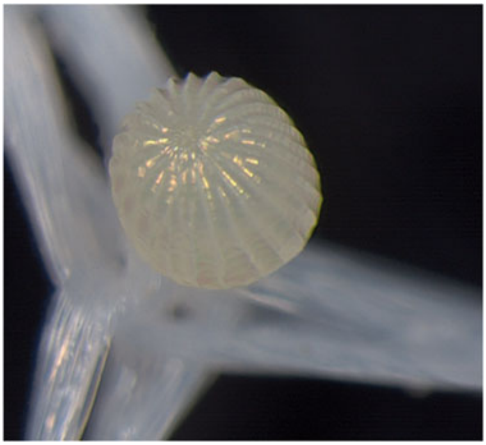

Fig. 7.2a Egg of *Helicoverpa armigera*. (Queiroz-Santos et al. 2017)

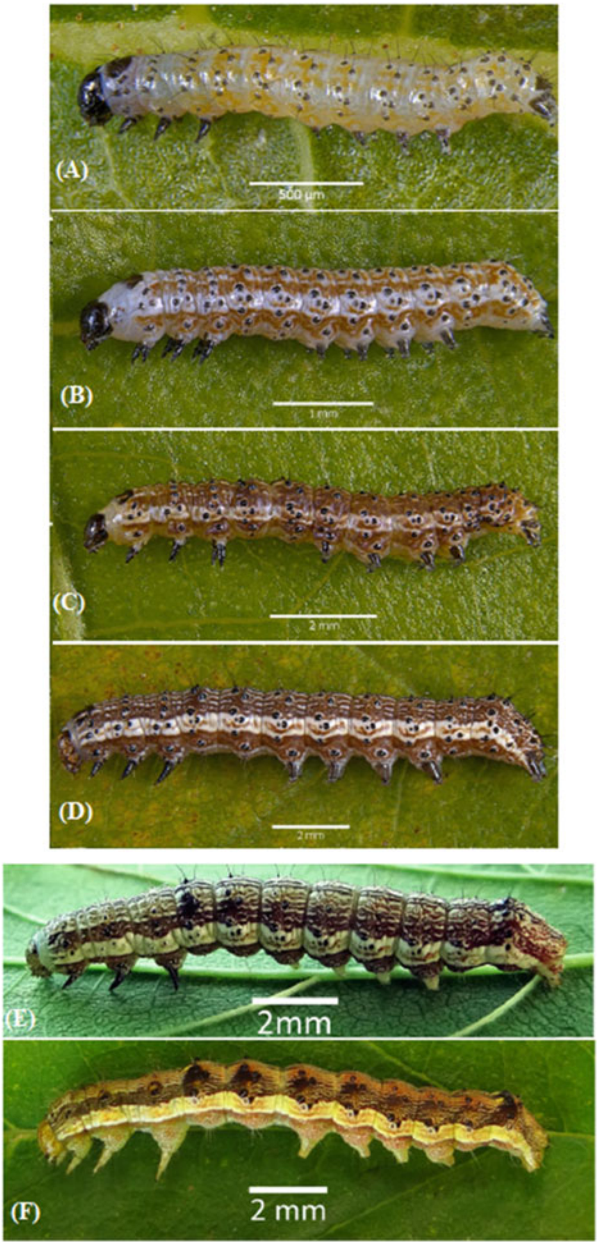

Fig. 7.2b Larval stages of *H. armigera*. (**A**) First instar, (**B**) Second instar, (**C**) Third instar, (**D**) Fourth instar, (**E**) Fifth instra, (**F**) Sixth instar. (Queiroz-Santos et al. 2017)

line bears a broad, light band with prominently visible spiracles. Most of these bands are wavy and wrinkled. The underside is uniformly rather pale (Fig. 7.2b).

7.4.3 Pupa

They are dark brown to mahogany brown and approximately 14–18 mm long. It possesses a smooth surface with rounded anterior and posterior ends. The posterior end bears two tapering parallel spines (Hardwick 1965; Fig. 7.2c).

7.4.4 Adult

As noctuid moths are, *H. armigera* too is a stout body moth with a wingspan of 3.5–4 cm. It possesses a broad thorax with a tapering body on either ends and a body length of approximately 14–18 mm. While the colour can be variable, male wings are usually in shades of greenish grey, while females are in tones of orangish brown. Forewings have seven to eight black spots on the margin along with a broad irregular more or less V-shaped transverse broad band. The hindwings are pale straw coloured with a broad dark brown border containing a paler roundish patch (Fig. 7.2d).

7.4.5 Life Cycle

The adults of *H. armigera* are nocturnal and emerge after sunset and are active at night. The adults have a promiscuous mating system with mating being elicited post calling behaviour and pheromone release. Females of *H. armigera* avoid random mating by using pheromone antagonist cis-11-hexadecenol against male courtship pheromones (Chang et al. 2017). Postmating females oviposit 300–3000 eggs during the next three weeks, which is their usual longevity during warm months. The eggs are laid scattered and singly on all parts of the plant except the underground parts. The neonate larvae feed extensively on various plant parts and grow up to a length of 40 mm. Thereafter, they leave the plants and enter the soil to pupate. Neonate larvae usually eat soft leaves, flowers or flower buds while moving about their feeding sites. Moulting occurs when larvae are about to develop into following larval stage. It takes the entire life cycle of 25–40 days in summers and 6–7 months in winters (Table 7.2). In the Middle East, pupal diapause in the winter months is quite common. Pupal diapause varies with environmental conditions, primarily

Fig. 7.2c Pupa of *Helicoverpa armigera*. (Queiroz-Santos et al. 2017)

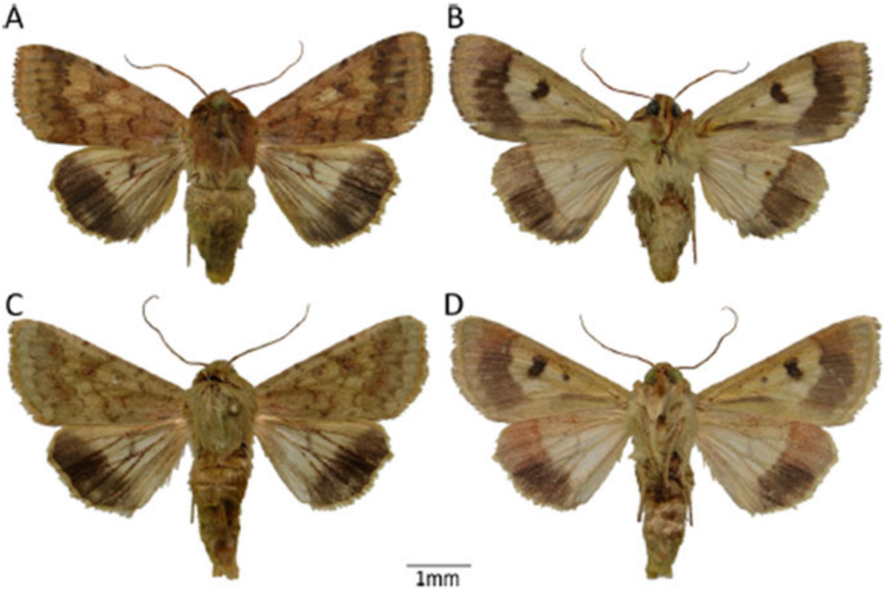

Fig. 7.2d Adults of *Helicoverpa armigera*. (**A**) Female dorsal view, (**B**) Female ventral view, (**C**) Male dorsal view, (**D**) Male ventral view. (Queiroz-Santos et al. 2017)

Table 7.2 Life cycle of *H. armigera* (Herald and Tayde 2018)

Stage	Developmental period (days)		
	Minimum	Maximum	Average ± SD
Incubation	3	4	3.50 ± 0.52
First instar	2	3	2.50 ± 0.52
Second instar	2	3	2.60 ± 0.51
Third instar	3	4	3.60 ± 0.51
Fourth instar	4	5	4.40 ± 0.52
Fifth instar	4	5	4.70 ± 0.48
Sixth instar	4	5	4.20 ± 0.42
Prepupa	1	3	2.10 ± 0.73
Pupa	13	15	13.80 ± 0.91
Male	8	10	8.90 ± 0.87
Female	10	14	11.90 ± 1.44

photoperiod and temperature (Hackett and Gatehouse 1982). The larval exposure to day lengths ranging from 11 to 14 hours/day and temperatures ranging from 15 °C to 23 °C induces facultative pupal diapause. Non-diapausing pupal stage varies from 6 to 30 days at temperatures ranging from 15 °C to 35 °C. Diapausing pupae are known to stay in this physiologically dormant state for durations ranging from several months to a year, with the later having been observed in the laboratory. In Sudan, *H. armigera* is known to undergo summer diapause due to extremely persistent hot and dry conditions (Hackett and Gatehouse 1982). *Helicoverpa* *armigera* is especially known for its ability to undergo wide-ranging facultative migration with distance travelled dependent on the prevailing environmental conditions. Evidence of migration has been found based on pollen traces in adults. During mass flight, it has been reported that under variable circumstances, altitudes may range from a few metres above crop canopy (Fitt 1989a; Drake and Fitt 1990; Riley et al. 1992) or at much higher altitudes (Feng et al. 2004, 2005a, b, 2009). The former is largely short pre-reproductive migration or non-migratory foraging movements (Drake and Fitt 1990; Drake and Gatehouse 1995; Drake et al. 1995; Drake and Reynolds 2012; Zhou et al. 2019), while the latter is long distance facultative migration and usually pre-reproductive (Coombs et al. 1993). The facultative nature of migration in *Helicoverpa* spp. is an evolutionarily strategic response to ephemeral inconsistent environmental conditions prevailing in agricultural areas (Dingle 2014).

7.5 Nature of Damage

The larval instars are the damage-causing stage in *H. armigera*. The young larval stages are primarily foliar feeders, feeding only on the chlorophyll and not the veins, therefore skeletonizing the leaves. The later larval stages cause the major damage to the crop, as they not only defoliate the plant but also shift their feeding sites to that of flowers and fruits. In the fruit-bearing pods, they bite holes through which they feed on all the seed and empty the pod completely. In cotton, they damage the cotton balls and squares. They bite holes into the base of balls and feed on the matter inside, turning them hollow; this results in the accumulation of moist frass at the base. Young larvae which are not strong enough to bore into the ball make small shallow marks, which attract microorganisms that induce rot. The later instars are, however, undoubtedly the most damage inducing as they damage larger fruits which are economically more devastating.

7.6 Management Measures

Saving the crops from infestations of *Helicoverpa armigera* is a herculean task. While there are a slew of control measures, cultural, chemical, biological, biotechnological, microbial and many more, across the world, chemical measures have been the most sought out after ones. The degree of loss due to *H. armigera* infestation depends on the number of larvae, their stage and also the temporality of the crop fruiting. While large larvae are responsible for most of the damage, they are virtually indestructible once they cross the third instar stage, which, therefore, indicates that control measures need to target eggs and stages younger than fourth instars (http://ipm.ucanr.edu/PMG/r114300511.html). Regular monitoring of pest populations is essential to manage pest populations. The use of light traps, pheromone traps and in situ assessments should be conducted intensively at all levels, viz. farm, village, block, regional and state levels. (http://agritech.tnau.ac.in/crop_protection/crop_

Table 7.3 Economic threshold (ET) and economic injury level (EIL) of *Helicoverpa armigera* on different crops

Crop	Economic threshold (ET)/economic injury level (EIL)	References
Chickpea	> 4 larvae/m^2	Odak and Thakur (1975)
	1.0 larva/m row	Singh and Reddy (1976)
	1.0 larva/m row	Patel (1994)
	1 larva/10 plants	Sekhar et al. (1994)
	0.6 larva/plant	Venkataiah et al. (1994)
	1.77–2.00 larvae/m row	Nath and Rai (1995)
	1.0 larva/m row	Whitman et al. (1995)
	0.81 larva/m row, 1.1 larva/m row	Zahid et al. (2008)
Pigeon pea	0.78–0.80 larvae/plant	Reddy et al. (2001)
Tomato	1.0 larva/plant	Cameron et al. (2001)
Cotton	19.86 larvae/100 plants	Alavil and Gholizadeh (2010)
Mung bean	1–3 larvae/m^2	Brier et al. (2010)
Peanuts	4 larvae/m^2	Brier et al. (2010)
Soybean	8 larvae/m^2	Rogers and Brier (2010)

prot_crop_insectpest%20_cotton.html). An economic threshold level of one egg/one larva per plant is considered most suitable for adoption of management practice. Different crops have different ETLs as mentioned in Table 7.3. We will, in this section, address the various control measures that are adopted in management of *H. armigera* populations.

7.6.1 Cultural Control

This control measures employ multiple cultural practices that have been standardized and passed down over generations in the farming community. Many agronomic practices such as early or late plantation, stubble cultivation, removal of crop residues, use of non-cropping seasons, removal of alternate hosts or breaking the synchronicity of their occurrence have been suggested for the management of *H. armigera* in various cropping systems (Fitt 1989a). These measures are effective by either of two methods, i.e. destruction of overwintering population or by providing a host free duration. Some of the cultural practices are listed below:

(i) Synchronized sowing with short-duration varieties.
(ii) Avoidance of monocropping. Planting of less preferred crops, such as green gram, black gram, soybean, castor and sorghum either in alternating rows or on field plot borders.

(iii) Trap crops can be planted to act as alternate and more attractive hosts than the crop plants. Alternatively, crops that act as reservoirs of natural enemies can also be planted nearby (Pearson 1958; Johnson et al. 1986). Spring trap crops have proven effective as an area-wide management tool for *Helicoverpa* population when it emerges from diapause in spring. Trap crop like okra when planted in the ratio of 25:1 row (cotton/bhendi) has proven to be effective in management of *Helicoverpa* populations in cotton crops. Red gram and marigold are also effective in trapping eggs of bollworms and killing eggs and young larvae (http://agropedia.iitk.ac.in/content/cultural-control-american-boll-worm).

(iv) Removal of alternate hosts from the vicinity of the agricultural crop can prevent the build-up of *Helicoverpa* and other insect pests.

(v) Crop residues can be a major source of pest carry-over to the next season. Their removal can help reduce increase in *Helicoverpa* in next season (Fitt and Forrester 1987).

(vi) Non-optimal use of nitrogenous fertilizers also favours multiplication of pest.

(vii) Excessive vegetative growth via increased watering can lead to increased areas for harbouring larvae.

(viii) Massive killing of overwintering pupae in the soil in temperate climates is an effective way of reducing the chances of *Helicoverpa* population in the coming seasons.

7.6.2 Host Plant Resistance

Planting of resistant crops is an important agronomic exercise for management of *H. armigera* populations and is especially effective in cotton, pigeon pea and chickpea. Crop variants possessing increased unpalatability, reduced preference or non-preference and increased tolerance to pests and infections have been exploited by breeders to produce pest-resistant varieties (Ashfaq et al. 2012; Sharma 2016; Ali et al. 2019; Kassi et al. 2019). ICRISAT, India, has specially been very successful at creating some resistant varieties of chickpea, groundnut and pigeon pea (Sharma 2016).

The introduction of the gene for the crystalline insecticidal protein of *Bacillus thuringiensis* into crops such as cotton and tomato has proven to be effective in reducing economic damage by *Helicoverpa* (Ai et al. 2019). However, what is worrisome is the increase resistance developing in *H. armigera* to even Bt crops (Alvi et al. 2012; Downes and Mahon 2012; Caprio et al. 2016; Dandan et al. 2019; Hazarika et al. 2019; Liu et al. 2019).

7.6.3 Chemical Control

While multifarious means have been used globally for the management of *H. armigera* populations, the most common one is the application of synthetic

Table 7.4 Insecticides for the control of *H. armigera*

Insecticide	Concentration
At early stages of square formation	
Acephate	75% SP 780 g/ha
Azadirachtin	0.03% EC 500 ml/ha
Carbaryl	10% DP 25 kg/ha
Chlorantraniliprole	18.5% SC 150 ml/ha
Chlorpyrifos	20% EC 1250 ml/ha
Diflubenzuron	25% WP 300 g/ha
Emamectin benzoate	5% SG 190–220 g/ha
Fipronil	5% SC 2000 ml/ha
Flubendiamide	20% WG 250 g/ha
Flubendiamide	39.35% SC 100–125 ml/ha
Indoxacarb	14.5% SC 500 ml/ha
Lufenuron	5.4% EC 600 ml/ha
Novaluron	10% EC 1000 ml/ha
NPV of *H. armigera*	0.43% AS 400–600 ml/ha
Profenofos	50% EC 1750–2500 ml/ha
Pyridalyl	10% EC 1500–2000 ml/ha
Spinosad	45.0% SC 165–220 ml/ha
Thiodicarb	75% WP 1000 g/ha
During bolling and maturation stage (1000 l of spray fluid/ha)	
Quinalphos	25 EC 2.0 l/ha
Carbaryl	50 WP 2.5 kg/ha
Pyraclofos	50 EC 1.5 l/ha

http://agritech.tnau.ac.in/crop_protection/crop_prot_crop_insectpest%20_cotton.html

insecticides such as organophosphates, synthetic pyrethroids and biorational compounds.

Application of insecticides is suggested largely at the early larval stages, because once the larvae enter into the pod/ball application of insecticides is largely ineffective. Some of the insecticides and their concentrations are described in Table 7.4.

While insecticide usage is the most prevalent one across the globe for managing *H. armigera* populations, it comes with unique set of problems. Of course, one of the most common problems with insecticide application is their generalist broad-spectrum activity, toxicity, biomagnification, bioaccumulation and persistence in nature. This causes long-term environmental harm as well as the reduction in populations of natural enemies in the vicinity, which in turn hampers the mechanism of natural control. Other than these common issues with insecticides in general, *H. armigera* represents a unique one, the issue of insecticide resistance. Across the globe, *H. armigera* populations are becoming resistant to a multitude of insecticides. *Helicoverpa armigera* has developed high-level resistance to multiple organochlorine, organophosphate and pyrethroid pesticides and may also show resistance to Bt in the near future. General advisory for insecticide usage is to discourage use of insecticides, primarily synthetic pyrethroids; prevent ad hoc application of insecticides; and use natural enemy and safe insecticides, such as endosulfan and phosalone. (http://agritech.tnau.ac.in/crop_protection/crop_prot_ crop_insectpest%20_cotton.html). Persistent indiscriminate insecticide usage has usually caused the entry into 'insecticide treadmill' where diminishing results are obtained in response to insecticide application.

Insecticide resistance in *H. armigera* has evolved in response primarily to the massive selection pressure of the reckless insecticide usage, especially synthetic pyrethroids (Ahmad et al. 2019; Chen et al. 2020a, b; Li et al. 2020; Sene et al. 2020). Inefficacy of pyrethroid application in controlling *Helicoverpa* populations has been reported from Australia, Thailand, Turkey, India, Indonesia and Pakistan (Karim 2000). Pyrethroid resistance in *H. armigera* has been attributed to three different pathways: detoxification by mixed-function oxidases, nerve insensitivity and delayed penetration (https://www.cabi.org/isc/datasheet/26757#topreventionAndControl; Carneiro et al. 2019; Tsakireli et al. 2019; Chen et al. 2020a, b). Organophosphate and carbamate resistance occurs due to decreased penetration, increased detoxification and/or insensitive AChE (Gunning et al. 1994, 1996b; Chitra and Reddy 1993). Pyrethroid resistance involves decreased penetration, increased hydrolysis, increased metabolism by monooxygenase and/or nerve insensitivity (Ahmad et al. 1989; Ahmad and McCaffery 1991; Gunning et al. 1995, 1996a; Phokela and Mehrotra 1989). Piperonyl butoxide and other synergists have been found to reduce or inhibit metabolic resistance and may act as major tools in alleviating the pre-developed insecticide resistance problem.

Insecticide resistance management strategies have evolved with the aim of either preventing it or containing it. These strategies largely involve strict rotation and time limitation in the use of pesticides and alternation of pesticide types. And while the migratory tendency of this noctuid moth does allow the dilution of local resistant populations, it also promotes the spread of resistance (Forrester et al. 1993).

In view of the tremendous obstacle that insecticide resistance has created globally in managing pest populations, the application of a well-thought insecticide resistance management systems (IRMS) has been proposed and encouraged. The IRMS aims to minimize the exposure to pesticides across consecutive generations, leading to reduced selection for pesticide resistance. This is proposed to be achieved by the following:

(i) Rotation of pesticide groups and alteration of pesticide usage.
(ii) Individual products and chemical groups to be avoided for persistent and long-term usages.
(iii) Early application of insecticides since even resistant larvae are susceptible till 4 days old (Daly 1988).
(iv) Product application windows to be decided based on pest life cycles and crop growth cycle.
(v) Mass culling of pupae.

7.6.4 Biological Control

Naturally, populations of predators and parasitoids are important in regulating *Helicoverpa* populations (King and Jackson 1989; Cherry et al. 2003; Allahyari et al. 2020; Table 7.5). Such natural enemies have the potential to maintain *Helicoverpa* populations at levels below the economic threshold level without the applications of insecticides. Effective usage of these natural enemies or biological control agents involves their conservation, augmentation and release.

The most common natural enemies of *Helicoverpa* in field crops are hemipterans (viz. spined predatory shield bug, assassin bug, damsel bug), predatory coleopterans (e.g. ladybird beetle, carabid beetle, soldier beetle), chryspoids, ants and spiders. Amongst parasitoids, most common ones under field conditions are small wasp species such as *Microplitis*, *Trichogramma* and *Telenomus*, relatively large parasitoid wasps (*Netelia*, *Heteropelma*, *Ichneumon*), flies (*Carcelia* and *Chaetopthalmus*), etc., (https://thebeatsheet.com .au/key-pests/helicoverpa-management/#bene; CABI 2020). Not only native predators but also exotic ones are helpful in biological control of *Helicoverpa*, with many having been introduced and established across the globe. Examples are *Trichogramma pretiosum* native to the USA well-established in Indonesia, *T. perkinsi* native to the USA established in South Africa, *Chelonus blackburni* and *Eucelatoria bryani* native to the USA established in India, *Bracon kirkpatricki* from Kenya established in India, *Cotesia marginiventris* from the USA established in Fiji, *Glabrobracon croceipes* from the USA established in New Zealand and *Cotesia kazak and Hyposoter didymator* from Europe established in Western Australia. None of these introductions appear to have had a significant beneficial impact (CABI 2020; Dassou et al. 2018).

Amongst pathogens, many infect and kill *Helicoverpa* larvae, including nuclear polyhedrosis virus (NPV) and fungal pathogens (*Metarhizium*, *Nomuraea* and *Beauveria*) (Eroglu et al. 2018; Sun et al. 2019; Khare and Gupta 2019; Khaskheli et al. 2019; Nawaz et al. 2019; Vidhate et al. 2019). The multiple issues faced due to insecticide applications have made microbial pesticides a favoured component of IPM strategies. Research on developing *Bacillus thuringiensis* and *Helicoverpa armigera* nuclear polyhedrosis virus (HaNPV) into commercially viable products is extensive globally. HaNPV in particular has been found to have some impact on *H. armigera* populations, although seldom reaching the levels necessary to achieve effective control. Ascovirus is also known to stunt larval development and is spread by wasp parasitoids (Chen et al. 2020a, b):

Some of the biocontrol practices commonly utilized in *Helicoverpa* management are as follows:

(i) Application of nuclear polyhedrosis virus (NPV) in evening hours at seventh and12th week after sowing.
(ii) Conservation and augmentation of natural predators and parasitoids.
(iii) Release of egg parasitoid, *Trichogramma* spp., at 6.25 cc/ha at 15 days interval three times from 45 days after sowing.

Table 7.5 List of natural enemies of *H. armigera*

Natural enemy	Life stages	Biological control in	Biological control on
Predators			
Acanthaspis pedestris			
Acridotheres tristis			
Andrallus spinidens			
Argiope bruennichi			
Brinckochrysa scelestes			
Bubulcus ibis	Larvae		
Camponotus sericeus			
Carabidae	Pupae		
Cataglyphis bicolor			
Cheilomenes sexmaculata			
Chrysopa	Eggs/ larvae		
Chrysopa congrua			
Chrysopa formosa		China; Shandong	Cotton
Chrysopa intima		China; Shandong	Cotton
Chrysopa pallens		Shandong	
Chrysopa phyllochroma			
Chrysoperla			
Chrysoperla carnea		Andhra Pradesh; China; Portugal; USSR; Tamil Nadu	Cotton
Chrysoperla sinica		Shandong	Cotton
Coccinella septempunctata		China; Shandong	Cotton
Coccinella transversalis		Australia; Queensland	*Citrus*
Coccinella undecimpunctata			
Deraeocoris punctulatus			
Diadegma fenestrale			
Dicyphus tamaninii			
Dohrniphora cornuta			
Dorylus helvolus			
Ectomocoris xaverei			
Edocla slateri			
Eocanthecona furcellata			
Erigonidium graminicolum		China; Shandong	Cotton

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Erigonidium graminicolum		China; Shandong	Cotton
Euborellia pallipes			
Eumenes maxillosus			
Geocoris	Larvae		
Geocoris amabilis			
Harmonia axyridis		China; Shandong	Cotton
Hemidactylus flaviviridis			
Hippodamia variegata		China; Shandong	Cotton
Laius venustus		Sudan	Cotton
Mallada boninensis			
Mallada desjardinsi			
Mepachymerus ensifer			
Microlestes discoidalis	Larvae	Sudan	Cotton
Micromus sjostedti			
Misumenops tricuspidatus		China; Shandong	Cotton
Monomorium indicum			
Nabis capsiformis		Sudan	Cotton
Nabis capsiformis	Larvae		
Nabis palifer			
Nabis sinoferus			
Nesidiocoris tenuis			
Oncocephalus annulipes			
Orius	Eggs/ larvae		
Orius albidipennis		Sudan	Cotton
Orius minutus		China	Cotton
Orius niger			
Orius similis			
Orius tantillus			
Orius thripoborus			
Paratrechina longicornis			
Pardosa astrigera			
Passer domesticus			
Podalonia tydei			

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Podisus maculiventris			
Praomys natalensis			
Propylea japonica	Eggs/ larvae	China; Shandong	Cotton
Rhynocoris fuscipes			
Rhynocoris kumari			
Solenopsis geminata			
Sterna aurantia			
Sycanus indagator		Mauritius	Tobacco; tomatoes; vegetables
Xysticus croceus		China; Shandong	Cotton
Xysticus mongolicus			
Zelus renardii			
Parasites			
Apanteles diparopsidis	Larvae		
Banchopsis ruficornis	Larvae		
Brachymeria lasus			
Brachymeria secundaria			
Bracon brevicornis	Larvae	Chad; South Africa	Cotton; field crops
Bracon gelechiae			
Bracon greeni	Larvae		
Bracon hebetor	Larvae	Israel; USSR	
Bracon kirkpatricki	Larvae	India	Cotton
Campoletis chlorideae	Larvae	Australia; China; India; India; Andhra Pradesh; Pakistan	*Amaranthus viridis*; *Cicer arietinum*; cotton; *Papaver hybridum*; polyphagous
Campoletis clavata			
Campoletis flavicincta			
Campoletis grioti			
Campoletis prinzi			
Campoletis rufegastor			
Campoletis sonorensis			
Campoplex collinus			
Carcelia cosmophilae	Larvae		
Carcelia illota	Larvae/ pupae		
Cardiochiles nigriceps	Larvae		

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Cardiochiles variegatus	Larvae		
Charops	Larvae		
Charops ater			
Charops bicolor		Pakistan	Tomatoes
Chelonus curvimaculatus	Larvae		
Chelonus formosanus			
Chelonus insularis	Larvae	South Africa	Fruit trees
Chelonus pilosulus	Larvae		
Compsilura concinnata	Larvae		
Copidosoma	Eggs		
Copidosoma obscurum			
Copidosoma truncatellum		Cape Verde	Beans
Cotesia flavipes	Larvae		
Cotesia glomeratus	Larvae		
Cotesia kazak	Larvae	Australia; Cape Verde; New Zealand; Queensland	Beans; *Cajanus cajan*; polyphagous; tomatoes
Cotesia marginiventris	Larvae	Australia; Cape Verde; Fiji; India	*Cajanus cajan*; polyphagous; tomatoes
Cotesia ruficrus	Larvae	Australia; Cape Verde	Beans; polyphagous
Drino imberbis	Larvae	Chad; Mauritius	Cotton; tobacco; tomatoes; vegetables
Eriborus argenteopilosus			
Eucelatoria bryani	Larvae	India	
Eucelatoria rubentis			
Euplectrus euplexiae			
Euplectrus laphygmae			
Eurytoma	Larvae/ pupae		
Exorista fallax	Larvae		
Exorista japonica	Larvae		
Exorista larvarum	Larvae	Egypt	Tomatoes
Exorista xanthaspis	Larvae	India; Gujarat	Sunflowers
Glabromicroplitis croceipes	Larvae	India; New Zealand	
Glyptapanteles africanus			

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Goniophthalmus halli	Larvae	Cape Verde; Chad; Kenya; Zimbabwe	*Cajanus cajan*; *Citrus*; cotton; tomatoes
Heteronychus arator			
Heteropelma scaposum	Larvae/ pupae		
Hyposoter didymator		Australia; Cape Verde	Beans; *Cajanus cajan*; polyphagous; tomatoes
Lespesia archippivora	Larvae	India	
Linnaemya longirostris	Larvae	Kenya	Tomatoes
Meteorus clytes	Larvae	Tanzania	Groundnuts
Meteorus ictericus	Larvae	Israel	
Meteorus laphygmarum	Larvae	Cameroon; Chad	Cotton
Meteorus pulchricornis	Larvae		
Microchelonus blackburni	Eggs/ larvae	Haryana; India; Maharashtra	*Cajanus cajan*; cotton
Microplitis	Larvae		
Microplitis demolitor	Larvae	Queensland	
Microplitis rufiventris	Larvae	Israel	
Nemoraea rubellana	Larvae		
Netelia testacea			
Pales coerulea	Larvae	Zimbabwe	*Citrus*
Pales pavida	Larvae		
Palexorista laxa	Larvae	Botswana	Sorghum
Palexorista quadrizonula	Larvae		
Palexorista solennis	Larvae		
Parania prima			
Peribaea mitis	Larvae	Sudan	*Trifolium*
Peribaea orbata	Larvae		
Pseudogonia rufifrons	Larvae		
Senometopia excisa	Larvae		
Senometopia kockiana	Larvae		
Sinophorus xanthostomus		New Zealand	
Spallanzania hebes	Larvae		
Steinernema carpocapsae			
Steinernema feltiae			

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Sturmiopsis inferens	Larvae		
Tachina praeceps	Larvae		
Telenomus	Eggs		
Telenomus busseolae			
Telenomus remus	Eggs	Cape Verde	*Cajanus cajan*; tomatoes
Telenomus ullyetti	Eggs		
Temelucha philippinensis			
Tetrastichus howardi			
Tetrastichus israeli			
Trichogramma	Eggs		
Trichogramma achaeae	Eggs		
Trichogramma bactriana	Eggs		
Trichogramma bourarachae	Eggs		
Trichogramma brasiliense	Eggs	India; Karnataka	
Trichogramma brassicae	Eggs		
Trichogramma chilonis	Eggs	Cape Verde; China; India; India; Gujarat; South Africa; Shandong	Beans; cotton; field crops; *Medicago sativa*; potatoes
Trichogramma chilotraeae	Eggs		
Trichogramma closterae	Eggs	China	Cotton
Trichogramma cordubensis	Eggs	Spain	Cotton
Trichogramma dendrolimi	Eggs	China; Shanxi	Cotton
Trichogramma evanescens	Eggs	Spain; Uzbekistan	Cotton
Trichogramma exiguum	Eggs	India	
Trichogramma fasciatum	Eggs	South Africa	Field crops
Trichogramma minutum	Eggs	India	
Trichogramma nubilale	Eggs		
Trichogramma perkinsi	Eggs	India; South Africa	Field crops; polyphagous
Trichogramma pintoi	Eggs	Spain	Cotton

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Trichogramma pretiosum	Eggs	India; Indonesia; South Africa; Karnataka	
Trichogramma rhenanum	Eggs		
Trichogramma semblidis	Eggs		
Trichogramma semifumatum	Eggs	South Africa	Field crops
Trichogramma sericini	Eggs		
Trichogramma urquijoi	Eggs	Spain	Cotton
Trichogrammatoidea	Eggs		
Trichogrammatoidea armigera	Eggs	Cape Verde	*Cajanus cajan*; tomatoes
Trichogrammatoidea australicum	Eggs		
Trichogrammatoidea bactrae	Eggs		
Trichogrammatoidea cojuangcoi			
Trichogrammatoidea lutea	Eggs		
Trichogrammatoidea nana	Eggs		
Trichospilus pupivora			
Winthemia lateralis	Larvae		
Xanthopimpla punctata			
Xanthopimpla stemmator			
Pathogen			
Bacillus cereus	Larvae		
Bacillus thuringiensis	Larvae	Sudan; Haryana; Shandong; Uttar Pradesh; Tamil Nadu; Delhi	
Bacillus thuringiensis aizawai	Larvae		
Bacillus thuringiensis entomocidus	Larvae		
Bacillus thuringiensis galleriae	Larvae		
Bacillus thuringiensis kenyae	Larvae		

(continued)

Table 7.5 (continued)

Natural enemy	Life stages	Biological control in	Biological control on
Bacillus thuringiensis kurstaki	Larvae		
Bacillus thuringiensis shandongiensis	Larvae		
Bacillus thuringiensis subsp. dendrolimus	Larvae		
Bacillus thuringiensis thuringiensis	Larvae		
Baculovirus heliothis		Taiwan	
Beauveria bassiana			
cytoplasmic polyhedrosis viruses	Larvae		
Granulosis virus			
Helicoverpa armigera nuclear polyhedrosis virus	Larvae		
Heliothis nucleopolyhedrosis virus			
Metarhizium anisopliae			
Nomuraea rileyi	Larvae		
Nosema furnacalis			
Nosema liturae			
Nosema medinalis			
Nosema pyrausta			
Nucleopolyhedrosis virus			
Serratia marcescens			
Vairimorpha necatrix			
Virus-like particles			

Sourced from CABI 2020

(iv) Use of parasitoid *Chelonus blackburni* and predator *Chrysoperla carnea* at 1,00,000/ha at sixth, 13th and 14th week after sowing.

7.6.5 Integrated Management

While each of the methods above plays a role in managing pest populations to some extent, it is the integration of various practices which has time and again proven to effectively manage pest populations.

7.6.5.1 Early Warning Systems

The migratory behaviour of *H. armigera* and its importance in the spread and bioinvasion of new areas suggest that installing means of monitoring their population and their movement can serve as an early warning system for nearby areas. Studies on radar, backtracking and other monitoring methods have revealed uncertainty and difficulty in predictability of their location and future movement. While trapping methods, especially that via light and pheromone traps, show clear abundance patterns across the Indian subcontinent (Srivastava et al. 1992), no strong relationships have been observed between trap catch and future egg or larval populations. Thus, as far as monitoring is concerned, trapping *H. armigera* is only useful as a qualitative measure which may indicate the initiation of an infestation or migratory tendencies, but not the directional nature or degree of movement the pests are likely to undergo.

7.6.5.2 Modelling

Models are theoretical or computational devices which are aimed with simulation of natural processes and improve their predictability. Models made post monitoring of extensive population growth data in relation to abiotic and biotic factors can be used to predict the time taken to reach economic threshold level and the efficacy of the measures deployed. All these predictions are of course subject to the limitations of each model. Models for the management of *H. armigera* have been used for the cotton crop in Australia and other *Helicoverpa* species in the USA. The SIRATAC system (a decision support system for cotton management) made by CSIRO and New South Wales Department of Agriculture, Australia, and other much improved models such as Helicoverpa Armigera and Punctigera Simulation (HEAPS) pay special attention to the biological measures such as adult movement and also the type of crop as well as non-crops in the vicinity. These models are able to provide personalized temporal specific pesticide application regimes (Zalucki et al. 1986; Dillon and Fitt 1990). However, these models till now have only been successful in the crop ecosystem because of the large scale of its plantation and also its high economic worth, which makes acceptance of expensive model-based directions much palatable.

7.6.5.3 Field Monitoring and Economic Threshold Levels

The ascertaining and utilization of economic thresholds is essential to the formulation of an IPM programme. Continuous intensive monitoring of pest populations in the files is essential in determining the threshold, its crossing and degree of control measures. However, it is very difficult to obtain a clear correlation for pest management as there are usually multiple external factors, not only environmental but also socio-economic, that may influence the management efficacy. Egg numbers have usually worked as an effective baseline for control decisions for IPM of cotton in Malawi and Zimbabwe since the early 1960s, with egg per two plants in twice weekly counts considered enough to initiate spraying measures (Matthews and Tunstall 1968). In Sudan Gezira, over two eggs or larvae per 18 plants (Haggis

1982) and, in Australia, two eggs per metre row (Wilson 1981) are used as thresholds.

Adult moth count via trapping at best works as supplementary information to egg and larval count while making decisions on application of control measures (e.g. Rothschild et al. 1982).

7.6.5.4 Whole-Farm or Regional Approach

A farming system that provides multiple food options throughout the year for *Helicoverpa* to feed upon is not likely to be able to effectively control or manage their populations, and the likelihood of large-scale losses is extremely high. Thus, it is essential not only to look at a particular field in a season but also to look at annual sowing regimes in ones field as well as neighbouring fields for a considerable distance. Such an area-wide management approach requires high levels of communication and cooperation between farmers, consultants and research/extension personnel.

The integration of the above-discussed approaches remains the best way to deal with the problem of *H. armigera*. Attempts have been made to develop integrated management approach for *H. armigera* using host plant resistance (Naseri et al. 2009, 2010; Soleimannejad et al. 2010; Fathipour and Naseri 2011) including transgenic Bt crops (Shelton et al. 2002), biological control (predators and parasitoids) (Abdi-Bastami et al. 2011), interference methods including sex pheromones (Reddy and Manjunatha 2000), biopesticides (especially commercial formulations of *Bacillus thuringiensis*) (Liao et al. 2002), cultural practices (including appropriate crop rotations, trap crops, planting date and habitat complexity) (Jallow et al. 2004) and selective insecticides (Rafiee-Dastjerdi et al. 2008).

7.7 Conclusions

The common trend towards reducing the application of synthetic insecticides for pest management has enhanced the interest in integrated pest management (IPM) programmes in general with those for *H. armigera* not being an exception. We need to develop a wide range of control measures and integrate them seamlessly into IPM systems in general and in particular for long-term management of *H. armigera*. These programmes can only be effective when they have been integrated with large-scale ecological data and related population dynamics data. Only then will it be possible to forecast the occurrence and the degree of infestation of *H. armigera* and the IPM to be applied. *Helicoverpa armigera* has proven to be a formidable pest and will require global will and unity to stop it in its tracks.

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Fall Armyworm (*Spodoptera frugiperda*)

8

Sharanabasappa S. Deshmukh, B. M. Prasanna,
C. M. Kalleshwaraswamy, Jagdish Jaba, and Bhagirath Choudhary

Contents

Abstract

The fall armyworm (FAW), *Spodoptera frugiperda* (J. E. Smith) (Lepidoptera: Noctuidae), originated from America but is reported recently from Africa and the Asia-Pacific. FAW has caused huge international concern since its outbreak in

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S. S. Deshmukh (✉) · C. M. Kalleshwaraswamy
Department of Entomology, College of Agriculture, University of Agricultural and Horticultural Sciences, Shivamogga, Karnataka, India
e-mail: sharanu.deshmukh@gmail.com

B. M. Prasanna
International Maize and Wheat Improvement Centre (CIMMYT), Nairobi, Kenya
e-mail: b.m.prasanna@cgiar.org

J. Jaba
International Crops Research Institute for the Semi-Arid Tropics (ICRISAT), Hyderabad, Telangana, India
e-mail: jagdish@cgiar.org

B. Choudhary
South Asia Biotechnology Centre (SABC), New Delhi, India
e-mail: bhagirath@sabc.asia

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Africa since 2016 and in Asia since mid-2018. The chapter mainly reviews its global distribution, life cycle, identification characters, strains, host plants, nature of damage, economic damage, and integrated pest management strategies available. The pest completes its life cycle on maize in 30 days (in warm summer months); in cooler temperatures, it may extend up to 60–90 days. For effective management of fall armyworm, different tools, viz., cultural control, agronomic management, breeding for resistance, natural enemies, and eco-friendly insecticides, should be used in an integrated approach. As the insect is recently introduced to Africa and the Asia-Pacific, possible management strategies and future cases of action are discussed.

Keywords

Fall armyworm · Life history · Nature of damage · Natural enemies · Management

8.1 Introduction

Invasive species are the biggest threat to the environment and cause ecological and economic losses (Wilson 1992; Evans et al. 2016). In the United States alone, invasive species have estimated to cause US$120 billion loss annually (Pimentel et al. 2005). The fall armyworm (FAW), *Spodoptera frugiperda* (J. E. Smith) (Lepidoptera: Noctuidae), originated from the Americas (Luginbill 1928; Sparks 1979) with wide host plants that include maize, sorghum, millets, rice, sugarcane, soybean, vegetables, and cotton (Prowell et al. 2004; Bueno et al. 2010; Padhee and Prasanna 2019). Fall armyworm is highly migratory in nature and has high fecundity, wide range of host plants, and voracious feeding behavior, without diapause. These characteristics make the fall armyworm a major destructive crop insect pests. Due to variation in weather conditions in the Americas, seasonal and continental movement of this pest from Canada to Argentina is noticed (Mitchell et al. 1991; Nagoshi and Meagher 2004, 2008). In eastern region of the United States, moth migration from the northeast to the southeast was noticed annually (Nagoshi et al. 2017).

Outside the Americas, FAW was first reported in West Africa in January 2016 (Goergen et al. 2016) and has spread to more than 40 countries across Africa (Prasanna et al. 2018a). In May 2018, this highly invasive insect pest was noticed for the first time in India on the maize crop in the Shivamogga and Davanagere districts and Karnataka state (Sharanabasappa et al. 2018a) and subsequently reported by Ganiger et al. (2018) and Shylesha et al. (2018).

8.2 Diagnostic Features and Life History

8.2.1 Diagnostic Features

The later instar larvae (fourth to the sixth instars) are brownish black with three white dorsal lines and alight lateral line. On the dorsal side of the larva, black tubercles are found which bear spines. The frons has white inverted “Y” line on the head, and four dark warts in a square form on the dorsal surface of the eighth abdominal segment (Fig. 8.1) (Prasanna et al. 2018a). Forewing of male is gray brown with white triangular patch at the apical region and circular spot at the center of the wing (Fig. 8.2e), whereas female has uniformly grayish brown forewings mottled with dark brown spots (Fig. 8.2f). The hindwings of both male and female are silvery white with a dark border (Oliver and Chapin 1981; Prasanna et al. 2018a; Sharanabasappa et al. 2018b; Ganiger et al. 2018).

As there is no diapause reported in this pest, overlapping generations are noticed in a cropping period (Sharanabasappa et al. 2020a), the same as in Africa where continuous host plants are available in off-season irrigated crops (Prasanna et al. 2018a). However, in America, where cooler climate exists, fall armyworm cannot survive; hence, it migrates to warmer regions in the winter months. Under tropical climates of Asia and Africa, where there is a bimodal pattern of rainfall, pest may thrive throughout the year causing economic losses.

Although both vegetative and reproductive parts are damaged by the fall armyworm, feeding injury results in whitish patches and ragged and elongated holes. Increased feeding further in the whorl may even affect the development of tassel. Extensive leaf damage due to *S. frugiperda* may significantly reduce the photosynthetic area, which may result in stunted plants and reduction in grain yield. At the reproductive stage, the larvae may bore through the side or top of the earhead and start feeding on kernels at milky stage, affecting the quality of the grain and yield.

8.2.2 Life History

Fall armyworm takes about 30 days to complete its life cycle on maize in warm summer months; however, in cooler temperatures, it may extend up to 60–90 days (Prasanna et al. 2018a). Detailed information on various stages of the pest is presented below.

8.2.2.1 Egg

The female adult lays about 1000 eggs in clusters on below or above the leaf surface of the maize plant, at the base of the plant, and also in whorls. The eggs are ventrally flattened. Immediately after laying, the eggs are of light green in color (Fig. 8.2a) for a day and then turn to golden yellowish and finally to black color before hatching. The female covers a layer of scales on the egg mass with moldy appearance. Egg hatching may take from 2 to 3 days with an average of 2.50 days (Prasanna et al. 2018a; Sharanabasappa et al. 2018b).

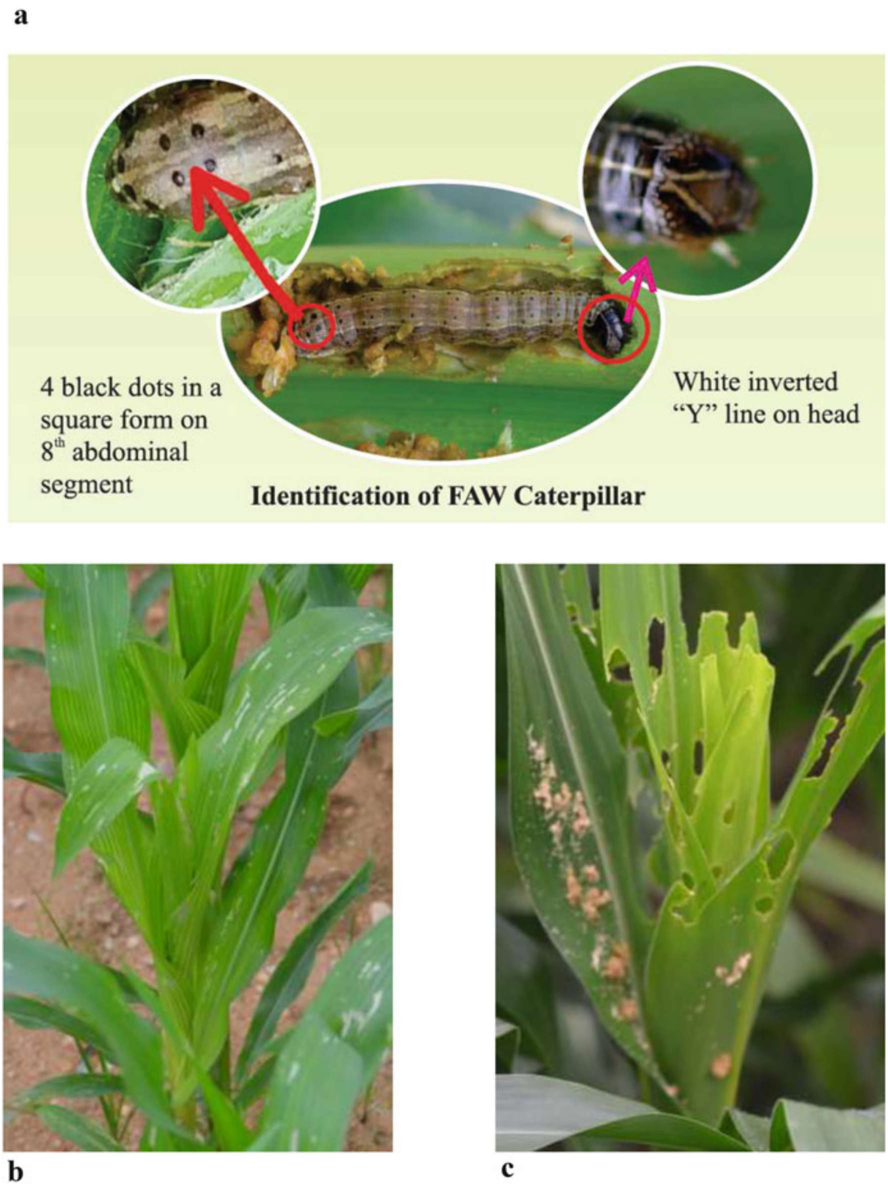

Fig. 8.1 (**a**) **Identification of fall armyworm caterpillar (Photos: Sharanabasappa)** (**b**) Scratches on the upper surface of the leaf due to feeding of the early instar larvae (**c**) Sawdust-like faecal matter found within the whorl and on upper leaves due to later instar larva

8.2.2.2 Larva

There are six larval instars with 14–19 days of larval duration, and color changes from instar to instar. First instars are green with a black head but it turns greenish

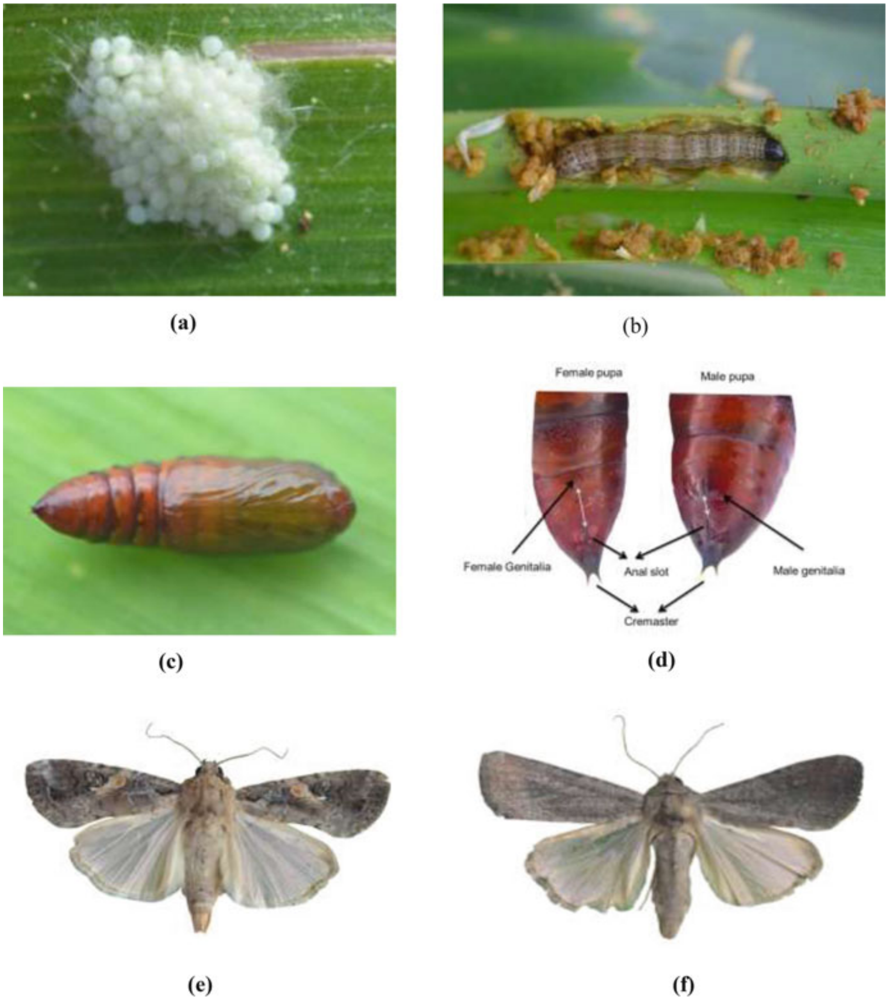

Fig. 8.2 Life stages of fall armyworm (**a**) Egg mass (**b**) Larva (**c**) Pupa (**d**) Pupal sexing in female and male (**e**) Adult Male (**f**) Adult Female. (Photos : Sharanabasppa)

brown during second instar. From third instar onward, larvae turn brown with three dorsal and lateral white lines (Fig. 8.2b) (Prasanna et al. 2018a; Sharanabasappa et al. 2019b, ICAR – IIMR 2019).

8.2.2.3 Pupa

Pre-pupa stops feeding and turns bright brown during pupal stages (Fig. 8.2c). Pupation takes place in the soil, and pupal period ranges from 9 to 12 days (Prasanna et al. 2018a; Sharanabasappa et al. 2019b). Pupal sexing can be done by looking at the genital opening. The distance from the genital opening to the anal slot can be used to distinguish the female and male pupa. The distance from the genital opening to the anal slot is more in female pupa than in the male pupa (Fig. 8.2d).

8.2.2.4 Adult

The adult longevity includes preoviposition, oviposition and post-oviposition periods ranged from 3 to 4, 2 to 3, and 4 to 5 days, respectively. In captivity, each female lays 835 to 1169 eggs with an average of about 1000 eggs. The total life cycle of male and female fall armyworm ranges from 32 to 43 and 34 to 46 days, respectively (Sharanabasappa et al. 2018b).

The female survives for 10.80 days with a range of 9–12 days compared to the male (8.20 days) with a range of 7–9 days. The average wingspan of female is 3.20 cm with a range of 3.00 to 3.4 cm, while it is 3.25 cm with a range of 3.00 to 3.50 cm in male (Sharanabasappa et al. 2018b).

Temperature and developmental rate of fall armyworm are linearly related between 18°C and 30°C and 26°C and 30°C, respectively. Studies demonstrated that 26°C and 30°C are the optimal range for egg, larval, and egg-to-adult development and lower larval maturity and the optimum temperature with the fastest larval development rate, and lowest mortality was at 30°C as reported by Du Plessis et al. (2020) (Table 8.1).

8.2.3 Strains of Fall Armyworm

FAW has two strains that are morphologically similar but differ in their host plant preference. The rice strain (R-strain) prefers to feed on rice, Bermuda grass, and other small grasses, whereas the corn strain (C-strain) prefers to feed on maize, sorghum, and other large grasses (Pashley et al. 1985; Pashley 1986; Pashley et al. 1987). These two strains do not have any clear biological attributes even though differences are evident in the whole genome, transcriptome, etc. (Gouin et al. 2017; Silva-Brandao et al. 2017). Mahadevaswamy et al. (2018) reported the presence of R-strain in population sampled from different parts of India using mtCOI gene, but this requires further validation. Maruthadurai and Ramesh (2020) reported the R-strain in the FAW population from Goa. It must be noted that the FAW population

Table 8.1 Duration of different life stages of FAW at different temperature regimes

	Temperatures (±1°C)				
Development stage	18	22	26	30	32
Egg (days)	6–7	4	3	2	2
Laval duration (days)	28–37	19–22	13–19	10–14	10–12
Pupal duration (days)	28–34	14–20	10–13	8–10	7–9
Egg to adult (days)	66–77	38–46	27–32	20–25	19–22
Larval mortality (%)	71	37	15	4	28

Source: Du Plessis et al. (2020)

in India preferentially damages maize, sorghum, and millets than rice. Hence, there is a confusion how mtCOI-detected R-strain is preferring to feed on rice in India.

In the Americas, the commonly used markers to identify morphologically indistinguishable C-strain and R-strain populations are mitochondrial *cytochrome oxidase subunit I* (*COI*) and nuclear *triosephosphate isomerase* (*Tpi*). Fall armyworm has two strains collections from Africa and India were reported to be R-strain detected by COI marker, although collections tested to date are from C-strain-preferred crops. When *Tpi* marker was used, >95% of the specimens were identified as C-strain. This indicates that *Tpi* marker is the ideal marker for identification of strains in Asian population. However, the presence of the R-strain in the Eastern Hemisphere needs constant watch, as it prefers to attack major host such as rice (Nagoshi et al. 2020).

The studies also suggest that fall armyworm from Africa and Asia has genetic similarity indicating invasion occurred from small number of population from the Western Hemisphere. The confusion of R-strain × C-strain might be due to inter-strain mating (Nagoshi et al. 2020).

Genetic evidence also suggests that FAW from Africa, India, Myanmar, and China shows that populations share a common and recent origin that derived from a small number of introductions (as few as one) from the Western Hemisphere. Nagoshi et al. (2020) provided two lines of evidence that suggest that a single strain predominates in the Eastern Hemisphere and that it is most likely the C-strain. Overall, they suggested that the FAW from Africa is behaving as expected for the C-strain, with the R-strain a minor presence or perhaps even absent. They also suggested that in Myanmar, China, India, and most of Africa, the *COI* strain marker is in disagreement with both *Tpi* and host plant. One way this could have occurred might be linkage between the mitochondrial *COI* marker and strain identity that was disrupted by inter-strain mating. Since mitochondria are maternally inherited, mating between an R-strain female and C-strain male would produce *COI*-RS hybrid daughters, which if they also mated with C-strain males would produce *COI*-RS progeny in a C-strain (including *TpiC*) background (Nagoshi et al. 2020).

Hybrid daughters may be produced with COI-RS strain male mates, as the mitochondria are maternally inherited. There is a need for comprehensive genetic analysis for invasive population to understand the strains and the host plants attacked. However, recent studies (Nagoshi et al. 2019, 2020) suggest the need for more extensive sampling of FAW on many more host plants across Asia for more reliable detection of host-associated differences.

8.3 Nature of Damage

Fall armyworm moths are nocturnal and hide during the daytime in the whorls of the maize plant. They are more active during the evening hours. Female moth lays eggs above/below the leaf surface of the maize plant, at the base of the plant, and also in whorls. Immediately after hatching, the neonate larvae secrete a silken thread and spread to the neighboring plants through wind. Early instars (1–3 instar stages) feed on the leaves, causing whitish patches appearing as "scratches" on the leaf surface (Fig. 8.1b). Grown-up caterpillars feed on leaf tissues resulting in ragged and elongated holes on leaves leading to sickly appearance. A very diagnosable symptom of attack is the presence of lumps of fecal matter in the whorls (Fig. 8.1c). Fall armyworm incidence starts when the crop is around 10–20 days after sowing. In early stage of the crop, 2–3 larvae are noticed feeding on the leaves, and in later stage of the crop, one or two later instar larvae per plant are noticed when the crop is around 30 to 40 days old. When two larvae are found in a single whorl, their feeding sites are different because in fall armyworm cannibalism is noticed. The early instar enters the cob through silk, whereas the later instar larva bores the husk and goes inside the cob and feeds on the kernels (Fig. 8.3a).

8.4 Global Distribution and Economic Damage

FAW has been found in 43 countries of Africa, 41countries in North America, 28 countries in Central America, and 32 countries in South America (Dively 2018). The pest is now widely prevalent in the Asia-Pacific region, including India, Yemen, Bangladesh, Sri Lanka, Thailand, Myanmar, China, Indonesia, Laos, Malaysia, Vietnam, Republic of Korea, Japan (FAO 2019), Nepal (Bajracharya et al. 2019), Indonesia (CABI 2019), and the Philippines (Navasero et al. 2019). Now the presence of this pest is reported in all states of India, except for a few northern states (Rakshit et al. 2019). Fall armyworm has spread to 26 provinces of China, the second biggest maize-producing country in the world after United States (Jiang et al. 2019). Recently, in February 2020, the pest has been reported in Australia (FAO 2020a; QGDAF 2020).

FAW is a highly invasive insect pest, with adult moth dispersal strongly influenced by wind and environmental conditions. FAW moths were reported to travel more than 100 km per day and nearly 500 km before egg laying, and they can move to newer places very quickly under favorable wind conditions. FAW is highly polyphagous; Montezano et al. (2018) reported a host range of 353 plant species from 76 plant families, principally Poaceae (106), Asteraceae (31), and Fabaceae (31). Despite such a broad host range, maize is undoubtedly the most widely preferred host by the pest. In Africa as well as in Asia, FAW damage has been mostly reported on maize, followed by a few other crops, like sorghum, millets, and vegetables. FAW has not adversely affected rice, despite its extensive cultivation in West Africa and many other sub-Saharan Africa countries (Rwomushana et al. 2018).

Yield losses due to FAW were estimated around 40 % in Honduras in Central America (Wyckhuys and O'Neil 2006) and 72 % in Argentina (Murúa et al. 2006). According to FAO, Brazil alone spends US$600 million annually on FAW management (Wild 2017). Abrahams et al. (2017) reported that FAW has the huge potential to cause 21% to 53% reduction in annual maize production (or US$2,481 to US $6,187 million economic damage) in 12 maize-producing African countries. In Africa, the maize losses were estimated at US$2.5 to 6 million due to fall armyworm in 2017 (Day et al. 2017).

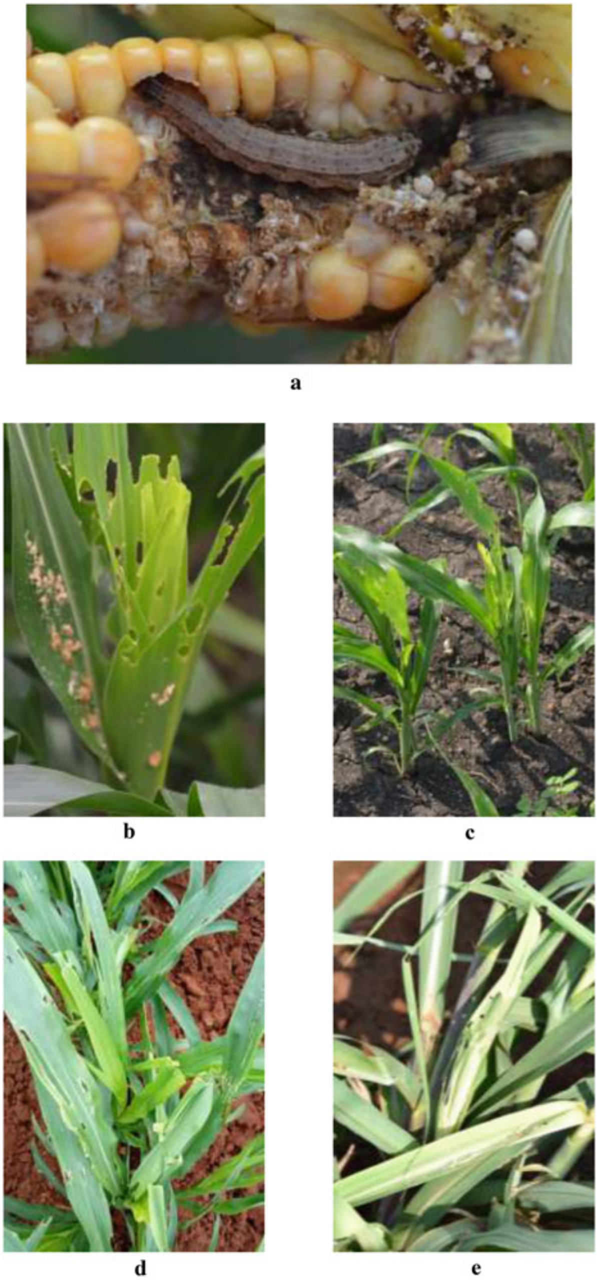

Fig. 8.3 (**a**) Fall armyworm feeding on cob (Photos: Sharanabasappa) (**b**) Maiza (**c**) Sorghum (**d**) Pearl millet (Phot: Jaba J) (**e**) Sugarcane

8.4.1 Crop Damage in India

Maize is the third most important cereal crop after rice and wheat, both in terms of area and production, registering maximum growth rate among food crops. In India, maize is grown in an area of 9.2 million hectares with a production of 27.82 million metric tonnes per year (FAO 2018). As reported by DMR (2012) and Chaudhary et al. (2012), the utilization pattern for maize in India includes mainly poultry feed (52%), human food (24%), animal feed (11%), and industrial processing (12%). The maize area affected due to fall armyworm in India in 2018–2019 was reported to be about 2.45 lakh hectares (Anonymous 2019). In India, it primarily feeds on maize (Fig. 8.3b), fodder maize, sweet corn, baby corn, and also other hosts, like sorghum (Fig. 8.3c), pearl millet (Fig. 8.3d), finger millet, sugar beet, and grasses, as reported by different workers (Table 8.2). In addition, FAW feeding was also reported on sugarcane (Fig. 8.3e) (Chormule et al. 2019) but not causing significant yield loss.

8.5 Integrated Pest Management (IPM)

The aim of IPM is to economically reduce the pest populations using suitable techniques and methods that minimize hazard to the environment, including people. IPM requires the farmers or farm advisors to have significant knowledge of agronomic and pest management approaches to implement an effective program based on local farming conditions (Prasanna et al. 2018a). The experiences so far in the Americas as well as Africa clearly show that there is no specific solution or magic bullet for effectively and sustainably controlling FAW. An IPM strategy based on science, inclusiveness, and balanced strategy is the need of the hour (Fig. 8.1). Emergency responses exclusively based on the use of synthetic pesticides have shown satisfactory results but need to be economically viable, to be used as per label claims, and must be safe for human health, biodiversity, and the environment (Fig. 8.4).

An effective IPM strategy for control of FAW will employ a toolbox approach, with different tools used in combination based on the cropping system, availability of technologies, and socioeconomic conditions of the farming communities. The IPM toolbox for FAW management could potentially include cultural control, agronomic management, host plant resistance, biological control, and environmentally safe synthetic biopesticides to protect the crops from economic injury while minimizing negative impacts on people, animals, and the environment (Prasanna et al. 2018a).

8.5.1 Monitoring, Surveillance, and Early Warning

Tracking and monitoring the spread of FAW across the country, region, and continent in a timely manner are critical if good decisions relating to control and management are to be made. Standard methodologies for field scouting and

Table 8.2 Host range of fall armyworm in India

S. no	Family	Scientific name	Common name	References
1	Poaceae	*Zea mays* L.	Maize	Sharanabasappa et al. (2018a), Ganiger et al. (2018), and Shylesha et al. (2018)
2	Poaceae	*Zea mays* L	Fodder maize	Maruthadurai and Ramesh (2020)
3	Poaceae	*Sorghum bicolor* L.	Sorghum	Sharanabasappa et al. (2018b), Jaba et al. (2019), Venkateswarlu et al. (2018), and ICAR – Indian Institute of Millets Research (2019)
4	Poaceae	*Eleusine coracana* (L.) Gaertn	Finger millet	ICAR – Indian Institute of Millets Research (2019), Jaba et al. (2019), and Venkateswarlu et al. (2018)
5	Poaceae	*Pennisetum glaucum* L. R. Br.	Pearl millet	ICAR – Indian Institute of Millets Research (2019), Jaba et al. (2019), and Venkateswarlu et al.(2018)
6	Poaceae	*Echinochloa frumentacea* Link	Barnyard millet	Roopika et al. (2020)
7	Poaceae	*Saccharum officinarum* L.	Sugarcane	Srikanth et al. (2018), Chormule et al. (2019), and Matti and Patil (2019)
8	Poaceae	*Brachiaria mutica* (Forssk.)	Para grass	Maruthadurai and Ramesh (2020)
9	Poaceae	*Megathyrsus maximus* (Jacq.)	Guinea grass	Maruthadurai and Ramesh (2020)
10	Amaranthaceae	*Amaranthus viridis* L.	Green amaranth	Maruthadurai and Ramesh (2020)
11	Amaranthaceae	*Beta vulgaris* subsp. *vulgaris* L.	Sugar beet	Shanthi et al. (2020)

collection of pheromone trap data are available (FAO 2020b; McGrath et al. 2018). In Africa, bucket traps and locally constructed traps are showing promise, while delta traps were found to capture fewer FAW moths. Determination of the most effective pheromone lure for FAW is less straight forward. While many pheromone lures attract about the same number of moths, the number of nontarget moths can vary significantly. In India, funnel-type pheromone traps are used for monitoring the FAW. Since the pest has invaded recently in India, studies on pheromone trap catches are going on. The maximum number of trap catches of FAW was recorded during 46th and 45th standard weeks in maize and sorghum crops (Jaba et al. 2019). There is an urgent need to develop FAW-species-specific pheromone lure with accurate pheromone blend ratio for effective pheromone lure catches.

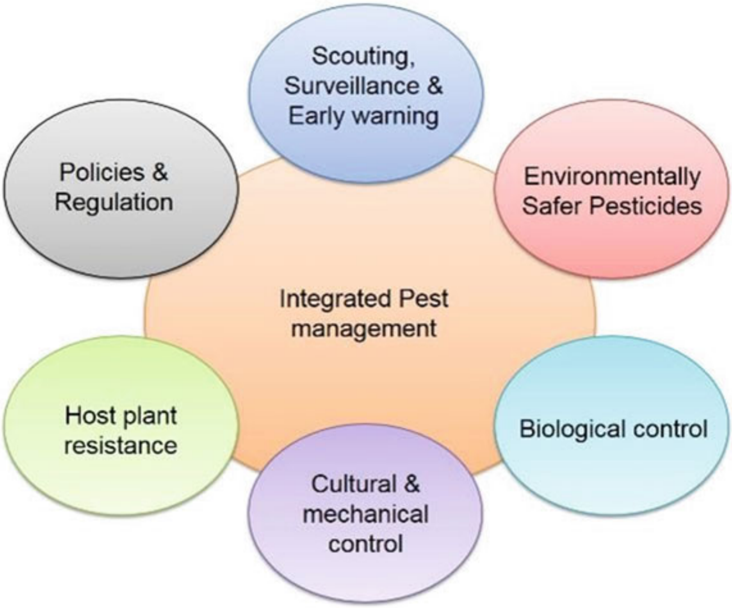

Fig. 8.4 Key components of an IPM-based strategy for FAW management

Given the visible nature of FAW, formal surveillance (including the use of appropriate pheromone traps) should be complemented by use of community-based crowd-sourcing techniques. Similarly, understanding and predicting dispersal patterns are essential if any early warning system is to be developed for effective control. Progress has been made on FAW environmental suitability models which can help to prioritize and improve scouting and trapping activities (Early et al. 2018). FAO established a Fall Armyworm Monitoring and Early Warning System (FAMEWS), which consists of the FAMEWS mobile app that was launched in March 2018 and a global platform that was established in July 2018.

8.5.2 Mechanical, Cultural, and Agroecological Management

The female moth lays up to 1500 eggs. These eggs are seen with naked eyes, and thus, farmers can identify and kill these egg masses with adequate training, as to prevent the caterpillars from damaging the crops. Manipulation of planting dates to ensure that the most susceptible stages of crop growth do not coincide with periods of peak moth activity is a well-known strategy to control lepidopteran pests. Planting early and adhering to regional planting calendar (avoiding late planting) will allow the maize crops to mature before buildup of high pest population. Tillage may destroy the pupae of lepidopteran pests (particularly in the case of FAW), as the pupae reside in the soil. Proper fertilization may also reduce plant damage by increasing plant defenses or increase it by making the plant more attractive to ovipositioning females. Intercrops may reduce crop infestation by lepidopteran pests through four possible ways: (i) by decreasing the movement of the caterpillars between maize plants, (ii) by decreasing the oviposition on maize plants, (iii) by emitting volatiles repelling ovipositioning females, and (iv) by hosting natural enemies.

The abundance of wide-ranging natural enemies of FAW (e.g., predatory arthropods, insectivorous birds, and/or bats) is a function of the availability of suitable habitat in the landscape, including hedgerows and non-crop habitat. Zero tillage and mulching may also create a favorable habitat for natural enemies and provide them with alternative prey. Push-pull strategy is another potential option for agroecological management of FAW (Midega et al. 2018).

8.5.3 Host Plant Resistance

Integrated pest management involving host plant resistance is a very important component against fall armyworm (Prasanna et al. 2018b).

Maize germplasm with naturally occurring or “native” genetic resistance to FAW was developed by CIMMYT in Mexico, where the pest was prevalent for several decades. Several research organizations such as CIMMYT (Mexico), EMBRAPA (Brazil), USDA-ARS (Mississippi), and universities in the United States led to the development of number of improved maize inbred lines with partial resistance to fall armyworm (Prasanna et al. 2018b). Similarly, sorghum and pearl millet germplasm also have native genetic resistance to sorghum stem borer; it may also be suitable for FAW developed by ICRISAT in India and Africa. Molecular biology tools now provide great potential for accelerating the development of new and promising varieties that could provide tolerance/resistance to fall armyworm and a host of other biotic stresses. Breeding for native genetic resistance to FAW is a medium- to long-term strategy and requires effective coordination and resources from the national partners and international organizations, like CIMMYT.

Transgenic/*Bt* maize producing endotoxins from the soil bacterium *Bacillus thuringiensis* (*Bt*) is one of the potential options for controlling a lepidopteran insect pest, like FAW. *Bt* maize technology is one of the most effective options to manage the FAW in both the United States and Brazil. In Africa, the TELA project has been testing *Bt* maize under confined field trials (CFTs) in six African countries to demonstrate the safety, efficacy, and yield benefits of the *Bt* maize under African conditions. Some of the African regulatory agencies have built capacity for science-based decision-making to address issues and societal concerns regarding *Bt* technology safety, effectiveness, and performance. Pyramiding transgenes with different novel modes of action (e.g., Cry + Vip genes) could be more effective and durable compared to single-gene deployment. Fast-tracked release of elite *Bt* maize varieties with FAW resistance could provide another powerful option in the IPM toolbox for FAW management. This, however, needs to go hand in hand with proper stewardship and insect resistance management to ensure durability of the technology.

8.5.4 Biological Control

Identification and use of natural enemies form basic component in IPM. Wherever necessary, inundative/augmentative release of well-validated biological enemies against FAW (e.g., *Trichogramma* sp. and *Telenomus* sp.; the egg parasitoids) should be taken up as a priority by public and private sector institutions in India. Several natural enemies have already been identified in African countries and in India, with reasonable levels of efficacy. In addition to biological control agents, bio-rational pesticides (like neem-based preparations) could also be potentially incorporated into the IPM-based strategies.

Survey carried out in the East African countries (Ethiopia, Kenya, and Tanzania) revealed four hymenopteran (*Cotesia icipe* Fernandez-Triana and Fiaboe (Braconidae), *Chelonus curvimaculatus* Cameron (Braconidae), *Coccygidium luteum* Brullé (Braconidae), *Charops ater* Szépligeti (Icheneumonidae)) and one dipteran (*Palexorista zonata* (Curran) (Tachinidae)) parasitoids (Sisay et al. 2018). With the exception of *C. curvimaculatus*, an egg-larval parasitoid, the rest are larval parasitoids. All these four species are native to Africa and not reported before from Africa or North and South America. Among these, one of the dominant larval parasitoids, *C. icipe*, with the high parasitism can be used for the management of fall armyworm (Sisay et al. 2018).

The two year surveys of fall armyworm natural enemies in maize and sorghum fields in Nigeria reported the egg parasitoids *Trichogrammatoidea* sp., *Trichogramma* sp., and *Telenomus* sp. and one egg-larval parasitoid *Chelonus* sp. and the other four larval parasitoids, viz., *Cotesia* sp., *Charops* sp., and unidentified ichneumonid and tachinid fly (Amadou et al. 2018).

The fungal pathogens like *Metarhizium anisopliae* and *Beauveria bassiana* are found effective against the eggs and second instar larvae of fall armyworm. Under laboratory conditions, *M. anisopliae* caused egg mortalities of 79.5%–87.0%, and *B. bassiana* recorded mortality of 30% to second instar larvae. The total mortality of eggs and early-stage larval mortality with *M. anisopliae* was as high as 96% with some fungal isolates (Akutse et al. 2019). Shylesha et al. (2018) recorded the egg parasitoids, viz., *Telenomus* sp., *Trichogramma* sp., *Glyptapanteles creatonoti* (Viereck), *Campoletis chlorideae* Uchida (Ichneumonidae), and *Cotesia ruficrus* on *S. frugiperda* larvae collected from the maize fields in Karnataka, India. Navik et al. (2019) recorded the natural parasitism (25.64%) by the *Trichogramma* sp. on fall armyworm eggs from Karnataka. Gupta et al. (2019) reported *Cotesia ruficrus* as an indeterminate larval-pupal ichneumonid parasitoid on fall armyworm, and the emergence of *C. ruficrus* adults from FAW larva is 11–29 wasps/larva. Sharanabasappa et al. (2019a) recorded larval parasitoids, namely, *Coccygidium melleum* (Roman), *Odontepyris* sp., and *Eriborus* sp. from Karnataka. In kharif 2019, the activity of two egg parasitoids, namely, *Trichogramma* sp. and *Telenomus remus*, was recorded from Shivamogga and Davanagere districts of Karnataka (Sharanabasappa et al. 2020a). Two predatory pentatomids, *Eocanthecona furcellata* and *Andrallus spinidens*, feeding on the larva of fall armyworm are reported by Shylesha and Sravika (2018). During the monsoon season (kharif) 2018, natural infestation of entomopathogenic fungi, *Metarhizium* (=*Nomuraea*) *rileyi* was noticed on *S. frugiperda* with its infection ranging from 1.87% to 18.30% (Mallapur et al. 2018) and 10% to 15 % (Sharanabasappa et al. 2019a). A comprehensive list of natural enemies reported from India is in Table 8.3.

8.5.5 Pesticide and Pesticide Risk Management

8.5.5.1 Pesticide

Insecticides are the necessary components in the insect pest management. The judicious use of insecticides is necessary for proper and effective pest management of the pest and least disturbance to the natural enemies and to the environment. The field efficacies of different insecticides against fall armyworm were investigated by many workers.

Another major issue with fall armyworm is the development of quick resistance to insecticides because of its behavioral and physiological factors (Yu 1991). Gutiérrez-Moreno et al. (2019) studied the field-evolved resistance of the fall armyworm to different insecticides. The LD_{50} values for flubendiamide, chlorantraniliprole, emamectin benzoate, and spinetoram against fall armyworm populations are collected from Mexico and Puerto Rico. The LD50 values of Puerto Rico are higher as compared to Mexico values because these insecticides are being used against FAW from many years in Puerto Rico.

Belay et al. (2012) studied the efficacy of different insecticides against FAW larvae under laboratory conditions. The insecticides like spinetoram, acephate, and thiodicarb recorded maximum ($\geq$60%) larval mortality as compared to lambda cyhalothrin and chlorantraniliprole. Similarly, Sisay et al. (2019) observed the insecticidal mortality to larvae of FAW under laboratory conditions showed that lambda cyhalothrin 5 EC recorded 77.8% larval mortality, and the next best were chlorantraniliprole plus lambda cyhalothrin 150 SC (62.2% mortality); spinetoram 120 SC recorded highest (61.1%) larval mortality and chlorantraniliprole 200 SC (60% mortality). At 48 and 72 h after treatment, spinetoram 120 SC caused the highest larval mortality of 96.7% and 100% larval mortality, respectively, whereas lambda cyhalothrin 5 EC recorded 96.7% mortality 48 h and 72 h after treatment applications. At present, the Central Insecticide Board and Registration Committee, India, recommended the ad hoc use of chlorantraniliprole 18.5 SC, thiamethoxam 12.6% + lambda cyhalothrin 9.5% ZC, emamectin benzoate 5 SG, and spinetoram 11.7 SC for fall armyworm management (DPPQS 2019). Mallapur et al. (2019) evaluated the efficacy of some insecticides both in laboratory and field experiments. The laboratory results revealed that spinetoram 11.7 SC and emamectin benzoate 5 SG were significantly superior over other treatments with 100% mortality at 60 hours after treatment. Under field conditions, spinetoram 11.7 SC, emamectin benzoate 5 SG, and spinosad 45 SC reduced the larval population to 98.13, 96.26, and 96.26 %, respectively, at 7 days after treatment. Muralimohan and Dileepkumar (2019) studied the efficacy of different insecticides as both sprays and poison baits (applied in whorls of infested plants). Spinetoram 11.7 SC, chlorantraniliprole 18.5

Table 8.3 List of natural enemies recorded on *Spodoptera frugiperda* in the maize fields in India

S. no.	Scientific name	Order: family	Nature of natural enemy	References
1	*Trichogramma* sp.	Hymenoptera: Trichogrammatidae	Egg parasitoid	Shylesha et al. (2018), Navik et al. (2019), and Sharanabasappa et al. (2020a)
2	*Telenomus remus* Nixon	Hymenoptera: Platygastridae	Egg parasitoid	Shylesha et al. (2018), ICAR-NBAIR (2019), and Patel et al. (2020)
3	*Chelonus formosanus* Sonan	Hymenoptera: Braconidae	Egg-larval parasitoid	Gupta et al. (2020b)
4	*Coccygidium melleum* (Roman)	Hymenoptera: Braconidae	Endo-larval parasitoid	Sharanabasappa et al. (2019a)
5	*Coccygidium luteum* (Roman)	Hymenoptera: Braconidae	Endo-larval parasitoid	Kannidi et al. (2020)
6	*Coccygidium transcaspicum* (Kokujev)	Hymenoptera: Braconidae	Endo-larval parasitoid	Gupta et al. (2020a)
7	*Campoletis chlorideae* Uchida	Hymenoptera: Ichneumonidae	Endo-larval parasitoid	Shylesha et al. (2018)
8	*Eriborus* sp.	Hymenoptera: Ichneumonidae	Endo-larval parasitoid	Sharanabasappa et al. (2019a)
9	*Exorista sorbillans* (Wiedemann)	Diptera: Tachinidae	Endo-larval parasitoid	Sharanabasappa et al. (2019a), Patel et al. (2020)
10	*Odontepyris* sp.	Hymenoptera: Bethylidae	Larval parasitoid	Sharanabasappa et al. (2019a)
11	*Cotesia ruficrus* (Haliday)	Hymenoptera: Ichneumonidae	Larval-pupal parasitoid	Shylesha et al. (2018) and Gupta et al. (2019)
12	*Forficula* sp.	Dermaptera: Forficulidae	Predator	Shylesha et al. (2018)
13	*Harmonia octomaculata* (Fabricius)	Coleoptera: Coccinellidae	Predator	Sharanabasappa et al. (2019a)

14	*Coccinella transversalis* Fabricius	Coleoptera: Coccinellidae	Predator	Sharanabasappa et al. (2019a)
15	*Eocanthecona furcellata* Wolff.	Hemiptera: Pentatomidae	Predator	Shylesha and Sravika (2018) and Sharanabasappa et al. (2020a)
16	*Andrallus spinidens* (Fabr.)	Hemiptera: Pentatomidae	Predator	Shylesha and Sravika (2018) and Sharanabasappa et al. (2020a)
17	*Spodoptera frugiperda* Nucleopolyhedrovirus	Baculoviridae	Entomopathogen	ICAR-NBAIR (2019) and Raghunandan et al. (2019)
18	*Metarhizium rileyi* (Farlow) Samson	Ascomycota: Clavicipitaceae	Entomopathogen	ICAR-NBAIR (2019), Mallapur et al. (2018), Sharanabasappa et al. (2019a, b)
19	*Bacillus thuringiensis*	Bacillales: Bacillaceae	Entomopathogen	ICAR-NBAIR (2019)
20	*Beauveria bassiana*	Hypocreales: Cordycipitaceae	Entomopathogen	ICAR-NBAIR (2019)
21	*Beauveria felina* (DC.) J.W. Carmich.	Hypocreales: Cordycipitaceae	Entomopathogen	Mohan et al. (2020)
22	*Heterorhabditis indica* Poinar, Karunakar, and David	Rhabditida: Heterorhabditidae	Entomopathogenic nematode	ICAR-NBAIR (2019)

SC, and novaluron 10 EC recorded highest larval mortality (93.53% to 96.76% reduction over untreated control) under laboratory conditions. Field studies suggested that bait application was as effective as foliar sprays involving same insecticide. The results indicated that greener molecules with a waiting period of < 3–5 days were very effective as baits, and cost of plant protection was substantially low when used as baits (up to 42% cost reduction). This finding provides an opportunity for the growers not only to reduce the cost of protection but also to make the food safe for consumption, particularly as fodder maize fed to the animals. Among the insecticides and biopesticides tested, thiodicarb 75% WP at 1g per lit, emamectin benzoate 5 SG at 0.5g per lit, and spinetoram11.7 SC at 0.5 ml per lit were found to be very effective against FAW, while pongamia oil at 6ml per lit of water was also found to be most effective among the biopesticides (Jaba et al. 2019). Similarly, Sharanabasappa et al. (2020b) found that chlorantraniliprole 18.5 SC at 37 g ai per ha, emamectin benzoate 5 SG at 12.5 g ai per ha, and spinetoram11.7 SC at 29.2 g ai per ha are suitable insecticides in managing the fall armyworm.

8.5.5.2 Pesticide Risk Management

Pesticides are classified based on hazards and risks which are used for the control of fall armyworm (Jepson et al. 2018). The suggested requirements for pesticide recommend the use of products that can be used with minimal protective clothing (PPE), which allows the reentry to the field, a day or less after spray or treatment. While there are plenty of crop protection products available across Africa and Asia, not all of them may be effective in controlling FAW. Products that farmers have been using range from chemicals, such as emamectin benzoate, chlorpyrifos, chlorantraniliprole, cyantraniliprole, emamectin benzoate, and lambda cyhalothrin, to biological, such as *Bt*, spinosad, spinetoram, and plant and biological extracts. Application of insecticides should be done late in the evening when the larvae are active (not hiding within the whorls); the larvae mostly feed in the early morning or at night when temperatures are not high and when there is no bright light.

Biological pesticides and natural enemies have been extensively studied, and a guide to candidate FAW biopesticides and biological control agents is published recently (Bateman et al. 2018). These include pesticides registered in 30 countries, 11 in Americas and 19 in African countries. Among the fifty biopesticide reported, twelve are found to be effective against FAW outside Africa, and these biopesticides are already registered to manage other pests in some African countries. A similar inventory needs to be drawn up soon in Asian countries.

8.6 Conclusions

Within a short span of 3–4 years, fall armyworm has spread to several countries across Africa and the Asia-Pacific, causing huge damage to the crops, especially maize, sorghum, and pearl millet in particular affecting the food surety, income, and subsistence of million farmers. Intensive research is required for developing economic thresholds, besides various aspects of the biology and behavior of fall armyworm in continents like Africa and Asia. Besides monitoring and surveillance, environmentally sustainable fall armyworm management requires effective integration of various approaches, including biological control, environmentally safe pesticides including biopesticides, host plant resistance, and agroecological management. In summary, fall armyworm poses a complex challenge and needs to be managed through well-coordinated, inter-institutional, and multidisciplinary efforts.

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Thrips

9

Bhupendra Kumar and Omkar

Contents

Abstract

Thrips are economically important insect pests across the global agroecosystems. They belong to order Thysanoptera. They are phytophagous, predaceous or mycophagous insect pests that inhabit a broad range of habitats. Although about 7700 species of thrips have been reported to date, hardly 1% of the species of this group damage the crops. Still their importance cannot be overestimated. Thrips have piercing and sucking types of mouthparts; as a result, they damage the crops either by feeding or by enormous oviposition. Since biocontrol methods using predaceous bugs and mites are largely unsuccessful, chemical insecticides are commonly used for managing thrips populations. However, entomopathogenic fungi are other potential biocontrol agents for certain thrips species. Nowadays, omic-based techniques and advanced computational systems are the powerful tools for crop protection. Assessing plant–thrips interactions,

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B. Kumar
Department of Zoology, Banaras Hindu University, Varanasi, India

Omkar (✉)
Department of Zoology, University of Lucknow, Lucknow, India

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exploring genetic diversity amongst thrips species and using resistant crop varieties may possibly assist in suppressing their populations below economic threshold in the near future. In this chapter, economic importance of thrips as insect pests and their management methods have been discussed so that they may not affect the global crop productivity.

Keywords

Thrips · Phytophagous pests · Biological control · Chemical control

9.1 Introduction

Thrips are amongst the economically important insect pests across the global agroecosystems. They are kept within hemipteroid assemblage along with Hemiptera, Psocoptera and Phthiraptera on taxonomic basis (Riley et al. 2018). They belong to order Thysanoptera (*thusanos*, a fringe; *pteron*, a wing; Tommasini and Maini 1995) and are either phytophagous, mycophagous or predaceous insects that occupy wide ranges of habitats, including tropical, subtropical and temperate geographical regions. Owing to their adaptive diversity, they have effectively exploited the diverse niches. They have successfully established themselves in a variety of plant formations and fungus-infested habitats like plant debris and bark of living and dead trees (see Ananthakrishnan 1993). Although about 7700 species of thrips have been reported to date, hardly 1% of the species damage the crops (Morse and Hoddle 2006). Still the importance of thrips cannot be overestimated. While some species are global pests, viz. *Frankliniella occidentalis* (Pergande) (Reitz 2009), *Thrips palmi* (Karny) and *Thrips tabaci* (Lindeman) (Murai and Loomans 2001; Diaz-Montano et al. 2011), others have regional distribution, like *Thrips hawaiiensis* (Morgan) (Murai 2001). So far, 14 thrips species are considered as vectors for tospoviruses (Rachana and Varatharajan 2018). Out of these, *F. occidentalis* transmits five species of tospoviruses, i.e. chrysanthemum stem necrosis virus, groundnut ring spot virus, impatiens necrotic spot virus, tomato chlorotic spot virus and tomato spotted wilt virus/groundnut bud necrosis virus (Riley et al. 2011a, b).

The herbivorous thrips feed on numerous plant species (Reitz 2009; Steenbergen et al. 2018) (Table 9.1). While some are polyphagous like those in genus *Frankliniella*, the others are oligophagous like *Megalurothrips sjostedti* (Trybom) and *Megalurothrips usitatus* (Bagnall) (Ngakou et al. 2008; Northfield et al. 2008; Tang et al. 2015). They have piercing and sucking types of mouthparts to feed on plant tissues. The life cycle includes the egg, actively feeding larvae, quiescent prepupae and pupae and adults that exhibit sexual dimorphism. Thrips have high fecundity and short developmental durations that result in their overlapping generations, high infestation rates and resistancy to different groups of chemical insecticides. These traits of thrips are responsible for enormous damages caused to agricultural crops, both by feeding and by oviposition (Bragard et al. 2019) (Fig. 9.1).

Table 9.1 List of thysanopteran pest species with their common names

Thysanopteran species	Common names
Actinothrips	
Adiheterothripidae	
Adraneothrips decorus Hood	
Aeolothrips astutus Priesner	
Aeolothrips bicolor Hinds	
Aeolothrips brevicomis Bagnall	
Aeolothrips fasciatus (L.)	
Aeolothrips intermedius Bagnall	
Aeolothrips melaleucus Haliday	
Aleurodothrips	
Allelothrips cameroni (Bagnall)	
Allelothrips cincticornis Bagnall	
Allothrips	
Anactinothrips	
Anaphothrips obscurus (Muller):	American grass thrips
Andrewarthaia kellyana (Bagnall)	
Androthrips	
Apterothrips apteris (Daniel)	
Aptinothrips rufus (Gmelin):	Grass thrips
Aptinothrips stylifer Trybom:	Grass thrips
Arrhenothrips ramakrishnae Hood	
Aurantothrips	
Bactrothrips brevitubus Takahashi	
Bactrothrips buffai (Karny)	
Bagnalliella yuccae (Hinds):	Yucca thrips
Caliothrips fasciatus (Pergande):	Bean thrips
[*Caliothrips fumipennis* (Bagnall and Cameron)]	
= *Caliothrips impurus* (Priesner)	
Caliothrips indicus (Bagnall)	
Caliothrips sudanensis (Bagnall and Cameron):	Cotton leaf thrips
Catinthrips vaccinophilus (Hood)	
Ceratothripoides cameroni (Priesner)	
Ceratothrips ericae (Haliday):	Heather thrips
[*Chaetanaphothrips clarus* (Moulton)]	
=*Chaetanaphothrips leeuweni* (Karny)	
Chaetanaphothrips orchidii (Moulton)	
Chaetanaphothrips signipennis (Bagnall):	Banana thrips
[*Chirothrips hoodi* Jacot-Guillarmod]	
= *Chirothrips ah* Girault	
Chirothrips manicatus Haliday:	Timothy thrips
Compsothrips	
Corynothrips stenopterus Williams:	Cassava thrips
Cryptothrips nigripes (Reuter)	

(continued)

Table 9.1 (continued)

Thysanopteran species	Common names
Cycadothrips chadwicki Mound:	Australian cycad thrips
Dactuliothrips	
Dendrothrips	
Diarthrothrips coffeae Williams:	Coffee thrips
Dichromothrips corbetti (Priesner):	Orchid thrips
Dichromothrips phalaenopsidis Sakimura:	Orchid thrips
Dichromothrips smithi (Zimmermann):	Orchid thrips
Drepanothrips reuteri Uzel:	Grape or vine thrips
Echinothrips americanus Morgan	
Elaphrothrips tuberculatus (Hood)	
Erotidothrips mirabilis Priesner	
Frankliniella bispinosa (Morgan)	
Frankliniella bondari Hood	
Frankliniella cephalica (Crawford)	
[*Frankliniella dampfi* Priesner]	
= *Frankliniella schultzei* (Trybom)	
Frankliniella fusca (Hinds):	Tobacco thrips
Frankliniella intonsa (Trybom):	Flower thrips
Frankliniella kelliae Sakimura	
Frankliniella lilivora Kurosawa	
Frankliniella minuta (Moulton)	
Frankliniella occidentalis (Pergande):	Western flower thrips
Frankliniella parvula Hood:	Banana flower thrips
Frankliniella schultzei (Trybom):	Common blossom or cotton bud thrips
Frankliniella tenuicornis (Uzel)	
Frankliniella tritici (Fitch):	Eastern flower thrips
Frankliniella vaccinii Morgan:	Blueberry thrips
Frankliniella williamsi Hood	
Franklinothrips megalops (Trybom)	
Franklinothrips tenuicornis Hood	
Franklinothrips vespiformis Crawford	
Gnophothrips fuscus (Morgan)	
Gynaikothrips ficorum (Marchal):	Cuban laurel thrips
Haplothrips aculeatus (Fabricius)	
Haplothrips chrysanthemi Priesner	
[*Haplothrips faurei* Hood]	
= *Haplothrips kurdjumovi* Karny	
[*Haplothrips floricola* Priesner]	
=*Haplothrips kurdjumovi* Karny	
Haplothrips gowdeyi (Franklin)	
Haplothrips hispanicus Priesner	
Haplothrips kurdjumovi Karny	
Haplothrips leucanthemi (Schrank)	

(continued)

Table 9.1 (continued)

Thysanopteran species	Common names
Haplothrips niger (Osborn)	
Haplothrips nigricornis (Bagnall)	
Haplothrips palaestinensis Priesner	
Haplothrips propinquus Bagnall	
Haplothrips sorghi Bagnall	
Haplothrips statices (Haliday)	
Haplothrips subtilissimus (Haliday)	
Haplothrips tritici (Kurdjumov)	
Haplothrips verbasci (Osborn):	Mullein thrips
Haplothrips victoriensis Bagnall	
Heliothrips haemorrhoidalis (Bouche):	Greenhouse thrips
Heliothrips sylvanus Faure	
Hercinothrips bicinctus (Bagnall):	Smilax thrips
Hercinothrips femoralis (Reuter):	and greenhouse or sugar beet thrips
Hoplandrothrips	
Hoplandrothrips marshalli Karny	
Hoplothrips karnyi (Hood)	
Hoplothrips pedicularius (Haliday)	
[*Isoneurothrips australis* Bagnall]	
= *Thrips australis* (Bagnall)	
Kakothrips pisivorus (Westwood):	Pea thrips
[*Kakothrips robustus* (Uzel)]:	Pea thrips
= *Kakothrips pisivorus* (Westwood)	
Karnyothrips	
Leptothrips mali (Fitch):	Black hunter
Limothrips cerealium Haliday:	Corn or Grain thrips
Limothrips denticornis Haliday:	Barley thrips
Liothrips oleae (Costa):	Olive thrips
Liothrips takahashii (Moulton)	
Liothrips vaneeckei Priesner:	Lily bulb thrips
Megalurothrips	
Megalurothrips distalis (Karny)	
Megalurothrips sjostedti (Trybom):	Bean flower thrips
Megalurothrips usitatus (Bagnall)	
Melanthrips arabs Priesner	
Melanthrips fuscus (Sulzer)	
Melanthrips pallidior Priesner	
Merothrips sp. (*brunneus* Ward)	
Microcephalothrips abdominalis (Crawford):	Composite thrips
Mycterothrips	
Mymarothrips	
[*Neoheegeria verbasci* (Osborn)]	
=*Haplothrips verbasci* (Osborn)	

(continued)

Table 9.1 (continued)

Thysanopteran species	Common names
Neohydatothrips variabilis (Beach)	
Neurisothrips fullawayi (Moulton)	
Odontothrips karnyi Priesner	
Odontothrips loti (Haliday):	Bird's-foot trefoil thrips
Oncothrips habrus Mound	
Oncothrips tepperi Karny	
Orothrips	
Parascolothrips priesneri Mound	
Parthenothrips dracaenae (Heeger):	Palm thrips
Phlaeothrips coriaceus Haliday	
Podothrips bicolor Seshadri and Ananthakrishnan	
Pseudodendrothrips mori (Niwa)	
Pygmaeolhrips eharaetis Shumsher Sigh.	
Pygmaeothrips columniceps Karny	
Retithrips syriacus (Mayet):	Castor thrips
Rhipiphorothrips cruentatus Hood:	Grapevine thrips
Scirtothrips aurantii Faure:	South African citrus thrips
Scirtothrips citri (Moulton):	Citrus thrips
Scirtothrips dorsalis Hood:	Assam or chilli thrips
Scirtothrips spinosus Faure	
Scolothrips brevipilis Priesner	
Scolothrips hoodi Priesner	
Scolothrips longicornis Priesner	
Scolothrips pallidus (Beach)	
Scolothrips rhagebianus Priesner	
Scolothrips sexmaculatus (Pergande):	Six-spotted thrips
Selenothrips rubrocinctus (Giard):	Cacao or red-banded thrips
Sericothrips bicornis (Karny)	
Sericothrips cingulatus Hinds	
Sericothrips langei Moulton:	Water lily thrips
Sericothrips staphylinus Haliday	
[*Sericothrips variabilis* (Beach)]	
=*Neohydatothrips variabilis* (Beach)	
Stenchaetothrips biformis (Bagnall):	Oriental rice thrips
Stenothrips graminum Uzel:	Oats thrips
Stichtothrips	
Stomatothrips	
Symphyothrips	
Synaptothrips distinctus (Bagnall)	
Synaptothrips gezinae (Faure)	
[*Taeniothrips alliorum* Priesner]	
=*Thrips alliorum* (Priesner)	
[*Taeniothrips annulatus* (Karny)]	

(continued)

Table 9.1 (continued)

Thysanopteran species	Common names
=*Thrips annulatus* (Karny)	
[*Taeniothrips atratus* (Haliday)]	
=*Thrips atratus* Haliday	
[*Taeniothrips discolor* (Haliday)]	
=*Thrips discolor* Haliday	
Taeniothrips eucharii (Whetzel)	
Taeniothrips inconsequens (Uzel):	Pear thrips
Taeniothrips orionis Treherne	
[*Taeniothrips meridionalis* Priesner]	
=*Thrips meridionalis* (Priesner)	
Taeniothrips picipes (Zetterstedt)	
[*Taeniothrips rhopalantennalis* Shumsher]	
=*Thrips flavus* Schrank	
[*Taeniothrips simplex* (Morison)]:	Gladiolus thrips
=*Thrips simplex* (Morison)	
[*Taeniothrips vaccinophilus* Hood]	
= *Catinathrips vaccinophilus* (Hood)	
[*Taeniothrips vulgatissimus* (Haliday)]	
= *Thrips vulgatissimus* Haliday	
Thrips alliorum (Priesner)	
Thrips angusticeps Uzel:	Cabbage, field or flax thrips
Thrips annulatus (Karny)	
Thrips atratus Haliday	
Thrips australis (Bagnall)	
Thrips calcaratus Uzel:	Basswood thrips
Thrips discolor (Haliday)	
Thrips flavus Schrank:	Honeysuckle thrips
Thrips florum Schmutz	
Thrips fuscipennis Haliday	
Thrips hawaiiensis (Morgan)	
Thrips imaginis Bagnall:	Apple blossom or plague thrips
Thrips major Uzel:	Rose thrips
Thrips nigropilosus Uzel:	Chrysanthemum thrips
Thrips obscuratus (Crawford):	New Zealand flower thrips
Thrips orientalis (Bagnall)	
Thrips palmi Karny	
Thrips parvispinus (Karny)	
Thrips physapus Linnaeus:	Dandelion thrips
Thrips pusillus Bagnall	
[*Thrips serratus* Kobus]:	Cane thrips
=*Fulmekiola serrata* (Kobus)	
Thrips setosus Moulton	
Thrips simplex (Morrison):	Gladiolus thrips

(continued)

Table 9.1 (continued)

Thysanopteran species	Common names
Thrips subnudula (Karny)	
Thrips tabaci Lindeman:	Onion or potato thrips
Thrips validus Uzel	
Thrips vulgatissimus Haliday	
Tiarothrips subramanii (Ramakrishna)	
Trichinothrips	
Varshneyia pasania (Mukaigawa)	
Veerabahuthrips	
Xylaplothrips	
Zaniothrips ricini Bhatti	

See Parker et al. (2013)

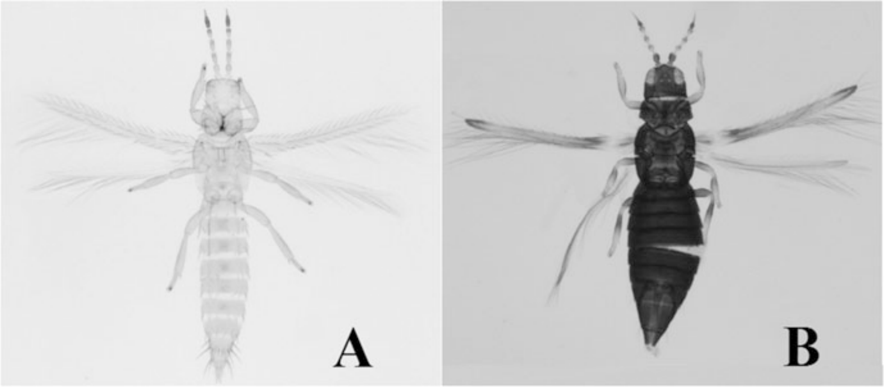

Fig. 9.1 (**a**) Male of *Frankliniella occidentalis*. (**b**) *Female of Caliothrips punctipennis*. (see Rachana and Varatharajan 2018)

Amongst the numerous management methods for thrips, use of chemical insecticides is the most common. However, resistance to insecticides is the major issue for thrips control. It is well documented that *Frankliniella occidentalis*, *T. tabaci* and *T. palmi* are resistant to numerous insecticides in several parts of the world (Herron et al. 2008; Bao et al. 2014; Wang et al. 2014). Although the use of resistant crop varieties may provide long-term solutions in controlling thrips and reducing insecticides use, physical control methods like coloured sticky traps may reduce the populations of thrips in greenhouses and open fields (Tang et al. 2016). In addition, use of natural enemies and entomopathogenic fungi is another method to control the increasing populations of thrips (Diaz-Montano et al. 2011).

Biocontrol of thrips using predatory bugs in genus *Orius* and mites in genera *Hypoaspis* and *Amblyseius* was previously found unsuccessful (Cox et al. 2006). However, entomopathogenic fungi have shown great potential in controlling certain species of thrips (Thungrabeab et al. 2006). Nowadays, 'omic'-based techniques and

Table 9.2 Major cosmopolitan thysanopteran pest species

Species	Host plant(s)	Biology
Thripidae		
Panchaetothripinae		
Heliothrips haemorrhoidalis	Polyphagous	Leaf and fruit living
Hercinothrips bicinctus	Polyphagous	Leaf and fruit living
Panhenothrips dracaenae	Polyphagous	Leaf and fruit living
Selenothrips rubrocinctus	Polyphagous	Leaf and fruit living
Thripinae		
Chaetanaphothrips clarus	Polyphagous	Leaf and fruit living
Chaetanaphothrips orchidii	Polyphagous	Leaf and fruit living
Chaetanaphothrips signipennis	Polyphagous	Leaf and fruit living
Dichromothrips phalaenopsidis	Orchidaceace	Leaf and fruit living
Frankliniella occidentalis	Polyphagous	Flower living, leaf and fruit living
Frankliniella schultzei	Polyphagous	Flower living
Microcephalothrips abdominalis	Polyphagous	Flower living
Thrips flavus	Polyphagous	Flower living
Thrips simplex	Iridaceae	Corm or bulb living, flower living
Thrips tabaci	Polyphagous	Flower living, leaf and fruit living
Phlaeothripidae		
Phlaeothripinae		
Gynaikothrips ficorum	*Ficus nitida*	Leaf and fruit living
Liothrips vaneeckei	Liliaceae, Orchidaceae	Corm or bulb living

See Vierbergen (1995)

advanced computational systems are providing powerful tools in crop protection. Understanding the complex interactions between host plants and thrips, assessing the genetic variations amongst thrips populations and using resistant crop varieties may possibly provide valuable information and new opportunities in minimizing agricultural losses by the thrips species (Mouden et al. 2017).

In this chapter, efforts are made to discuss the importance of thrips as insect pests of agricultural crops (Table 9.2). This chapter emphasizes on the general distribution and biology of thrips and how they attain the pest status. Attempts have been made to highlight the appropriate approaches in managing their populations below economic threshold levels so that the crop of major agroecosystems of the world may not be affected.

9.2 Systematic Position and Life History

Most thrips species are tiny and usually 1–2 mm long. The smallest species is about 0.5 mm, while the largest species is about 14 mm in length. They have elongated and dorsoventrally flattened bodies (Lewis 1973). The head has two large compound eyes and three ocelli between the two compound eyes. The antennae are 4–9-segmented. Right mandible is vestigial, but the left mandible is fully developed.

Table 9.3 Classification of order Thysanoptera

Order	Suborder	Family	Subfamily
Thysanoptera	Terebrantia	Aeolothripidae	
		Melanthripidae	
		Merothripidae	
		Stenurothripidae	
		Heterothripidae	
		Uzelothripidae	
		Hemithripidae	
		Thripidae	Panchaetothripinae
			Dendrothripinae
			Sericothripinae
			Thripinae
	Tubulifera	Phlaeothripidae	Phlaeothripinae
			Idolothripinae

See Mound and Morris (2007)

Two pairs of elongated wings are fringed with long cilia, and a bladder is located at the apex of each leg. The abdomen is ten-segmented (Lewis 1973).

Thrips have been placed taxonomically into two suborders, i.e. Terebrantia and Tubulifera, comprising 14 families. Of them, five families are known only from the fossils (Mound and Morris 2007; Mound 2013). Out of the remaining nine families, eight families belong to Terebrantia, and one family belongs to Tubulifera. Suborder Terebrantia comprises about 2000 species, whereas suborder Tubulifera includes about 3600 species globally (Tyagi and Kumar 2016). From India, a total of 739 species of thrips in 259 genera have been listed. Out of them, 309 species are from 116 genera belonging to suborder Terebrantia and 430 species from 143 genera belonging to suborder Tubulifera (Tyagi and Kumar 2016). The suborder Terebrantia is divided into eight families: Aeolothripidae (=Palaeothripidae), Melanthripidae, Merothripidae (=Jezzinothripidae), Stenurothripidae (=Adiheterothripidae), Thripidae, Uzelothripidae, Hemithripidae (=Fauriellidae) and Heterothripidae. In contrast, the suborder Tubulifera consists of only one family, Phlaeothripidae (Table 9.3).

Both Terebrantia and Tubulifera are differentiated from one another by numerous contrasting characters, like their skeletal morphology, wing structure (i.e. venation, chaetotaxy and coupling mechanism), structure and ontogeny of genitalia, the fine structure of sperm and their developmental biology. Forewings of adult Tubulifera have smooth surfaces without longitudinal veins, and the fringing cilia insert directly into the wing membrane. When adults are in resting position, their wings lie flat over one another on abdominal tergites held in place by one or two pairs of sigmoid setae (Nakahara 1991) (Fig. 9.2). Their tenth abdominal segment is tubular in shape, and the anus is terminal in position. The genital opening is located at the base of tube, and the female's ovipositor (being eversible) is chute-like in shape. Contrary to these morphological features, the forewing surface of adult Terebrantia is covered with

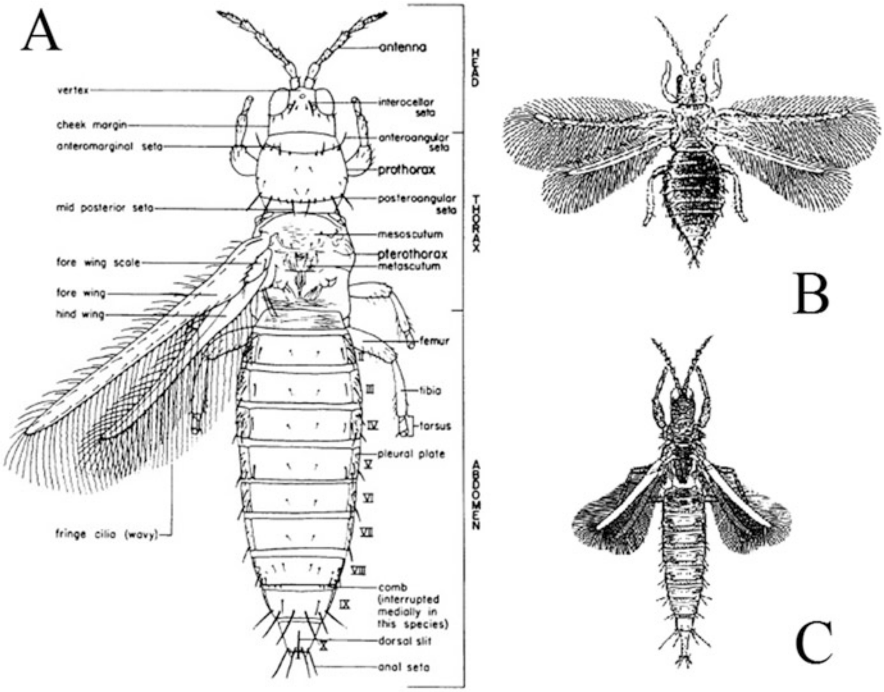

Fig. 9.2 (**a**) Morphology of Thysanoptera. (**b**) Adult of Thiripidae. (**c**) Adult of Phlaeothripidae. (see Nakahara 1991)

microtrichia, two longitudinal veins are visible, while the fringing cilia are inserted into sockets that are '8'-shaped. When adults of Terebrantia are at rest, their wings remain parallel to each other on the abdomen. Although the cilia of posterior margins remain in midline, the wing-holding mechanisms are different amongst species. The tenth abdominal segment is incomplete ventrally, but ovipositor comprises four saw-like blades for depositing eggs into plant tissues (Nakahara 1991).

The life cycle of thrips consists of the egg, actively feeding larval stages, inactive prepupal and pupal stages and an adult stage. The post-embryonic development lies in between hemimetabolous and holometabolous type. While the larvae resemble the adults (hemi-), the pupae undergo major dissolution of various imaginifugal structures (late L2 stage) and reformation of internal anatomical structures (holo-) including those of sensory and reproductive organs and the digestive tract (Lewis 1973; Moritz 1995). Such a metamorphosis in thrips is referred as 'remetaboly' (Moritz 1995) (Fig. 9.3). The incubation period and developmental durations of thrips are species-specific and depend upon the environmental conditions (Lowry et al. 1992). Eggs generally hatch after 2–26 days, depending upon the temperature and humidity. The plant-feeding larvae emerge out post hatching, and their development period ranges from 2 to 13 days (Lowry et al. 1992). Eddy and Clarke (1930) reported that at 24.7 °C, the first and second larval instars of onion thrips required 2.3

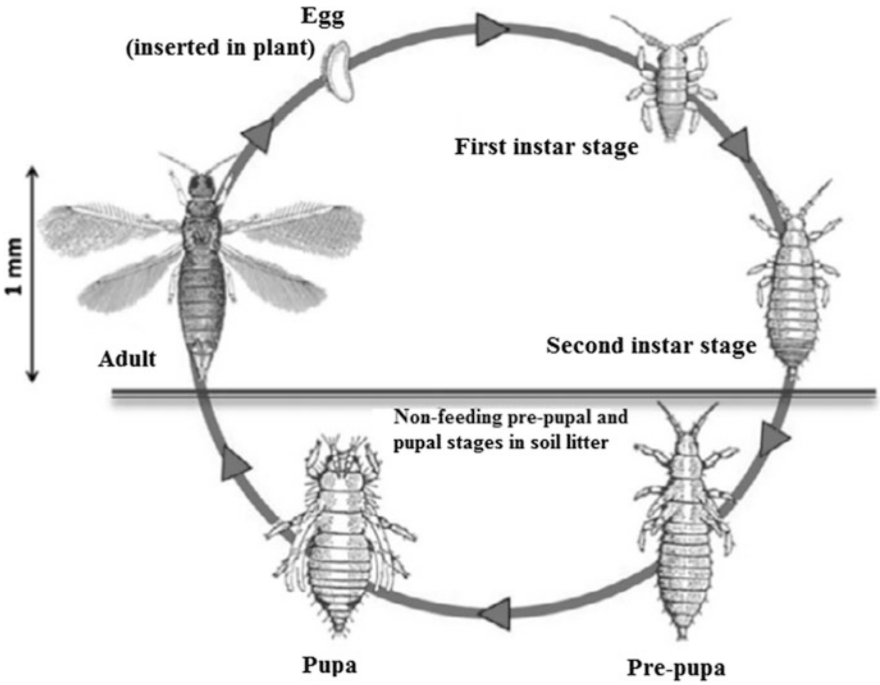

Fig. 9.3 Life cycle of *Thrips palmi* showing developmental stages. (see Bragard et al. 2019)

and 2.9 days for development. However, western flower thrips required 1.9, 2.3 and 1.1 days to complete the developmental durations of first instar and 9.8, 5.2 and 4.3 days to complete those of second instar at 15 °C, 20 °C and 30 °C, respectively (Lublinkhoff and Foster 1977).

The last larval stage is followed by a mobile but non-feeding prepupal stage. After 1–5 days post prepupal stage, the insect drops to the soil and enters the pupal stage. Adult thrips emerge from the soil after 1–10 days (Lublinkhoff and Foster 1977; Lowry et al. 1992). The onion thrips require 1–2 days and 4–7 days to complete the prepupal and pupal stages at 19 °C (MacGill 1927). However, the western flower thrips require 2.9, 2.2 and 1.4 days to complete their prepupal stage and 5.6, 2.9, and 1.6 days to complete pupal stage at 15 °C, 20 °C and 30 °C, respectively (Lublinkhoff and Foster 1977). The longevity of female onion thrips is 14.5 days and that of western flower thrips is 27.5 days at 30 °C, while it is 70.8 days at 15 °C for both the species. The onion thrips, tobacco thrips and western flower thrips are capable of reproducing sexually and asexually through parthenogenesis (Lowry et al. 1992). However, the soybean thrips can reproduce only sexually. Both the reproduction and development of thrips strongly depend upon the environmental conditions, especially temperature (Edelson and Magaro 1988). Although photoperiod does not induce diapause in many thrips species, adults exhibit temperature-induced reproductive quiescence (Jenser and Szenasi 2004). In temperate regions, cold weather delays the egg production, but breeding continues as the warm weather arrives during spring (Sites and Chambers 1990).

9.3 Commonly Found Thysanopteran Pest Species

At global level, the following thysanopteran species are considered as polyphagous pests and voracious feeders of agricultural crops.

9.3.1 *Frankliniella occidentalis* (Terebrantia; Thripidae: Thripinae)

The western flower thrips, *Frankliniella occidentalis*, is a thysanopteran species of Nearctic origin. It was first reported by Pergande (1895) in California on potato and apricot leaves. Lengthwise, the adult males of *F. occidentalis* measure 0.9–1.1 mm from the tip of antennae to the tip of the abdomen, while adult females measure 1.3–1.4 mm in length. Both the young larvae and adults have piercing and sucking type of mouthparts. They feed by piercing the leaf cells with their mandibles and ingesting cell contents through their feeding tubes formed by their maxillary stylets. The females bear a saw-like ovipositor with which they drill holes into parenchymal tissues of leaves, flowers and fruits and deposit their eggs of 0.25 × 0.50 mm in size (Brodsgaard 1989) (Fig. 9.4a).

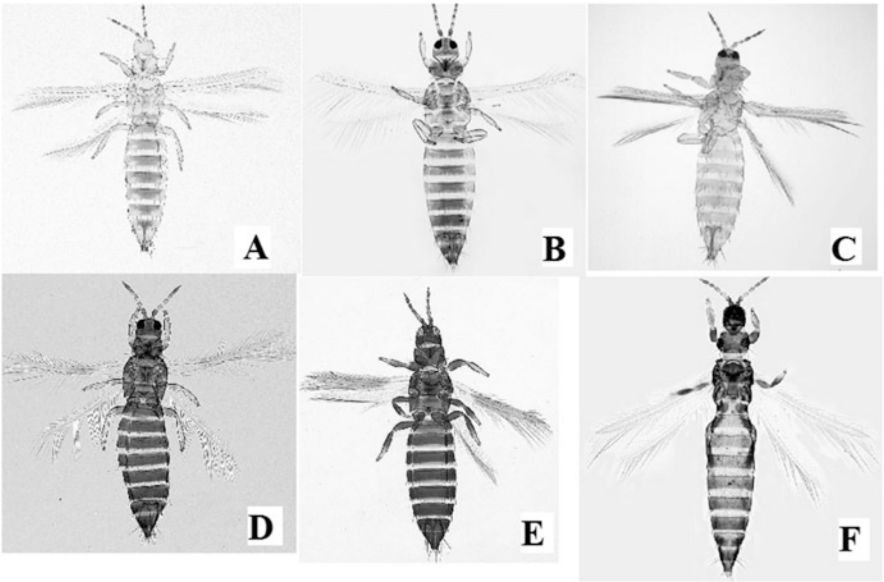

Fig. 9.4 (**a**) *Frankliniella occidentalis*, (**b**): *Thrips tabaci*, (**c**) *Thrips palmi*, (**d**) *Thrips fuscipennis*, (**e**) *Thrips simplex*, (**f**) *Thrips meridionalis*

Frankliniella occidentalis is a polymorphic pest species and displays three different colour forms (Sakimura 1962), viz. black, intermediate and pale coloured. The black- and pale-coloured forms have homozygous genotypes, while the intermediate form has a heterozygous genotype. Only diploid females feature all the three possible chromatic phenotypes, while haploid males are pale coloured. The post-embryonic development of *F. occidentalis* involves two larval stages, a prepupal and a pupal stage prior to the emergence of adults (Tommasini and Maini 1995). The newly hatched larvae are glassy white in colour. They start feeding immediately post emergence and become yellowish in colour as they mature. Morphologically, the larval instars are similar to adults, but the former are wingless and bear reddish eyes and few segmented antennae. The second instar larvae are smaller than the first instars; however, they are more active and highly voracious than the first instars (Tommasini and Maini 1995). The second instar larvae acquire a yellowish waxy colour, display positive geotaxis and negative phototaxis and move away from the plant towards the soil. At a depth of 1.5–2.0 cm below the soil, the larvae develop into the prepupal stage (Arzone et al. 1989). The prepupae are whitish in colour and are immobile unless they are disturbed. The pupal stage remains within the soil, develops longer antennae which face backwards towards the abdomen, becomes immobile and stops feeding. The adults emerge after the last moult and acquire a whitish colour which becomes darker within 48 h of moulting (Brodsgaard 1989). Soon after emergence, the adults begin to feed voraciously. The duration of development from eggs to adults is closely related to environmental conditions and is negatively affected by the change in temperature (Tommasini and Maini 1995; Stacey and Fellowes 2002).

Reproduction in *F. occidentalis* is of facultative parthenogenesis type, i.e. partly bisexual and partly parthenogenetic. Parthenogenetic reproduction is always arrhenotokous. The unfertilized females lay eggs that develop into males, while the eggs given by fertilized females develop into females (Brodsgaard 1989). Both larvae and adults are thigmotactic and are attracted to buds and flowers (like chrysanthemum) that provide protection from natural enemies and allow insect to go deep inside the plant. The adults overwinter on plants, like *Amaranthus* L., *Solanum nigrum* L., *Chenopodium* L. and *Heliotropium europaeum* L. (Marullo 1991), and at the end of winters, the females develop mature eggs (Mouden et al. 2017).

Adults of *F. occidentalis* are found on major crops, viz. *Rosa* L. spp. (rose), *Dianthus* L. spp. (carnation), *Gypsophila* L. spp., *Limonium* Miller spp., *Aster* L. spp., *Chrysanthemum* L. spp., *Ruscus* L. spp., *Solidaster* spp., *Impatiens* L. spp. (wax flower) (Gokkes 1991), *Anagallis arvense* L., *Senecio vulgaris* L., *Papaver rhoeas* L. (Del Bene and Gargani 1989) *Amaranthus*, *Heliotropium europium*, *Chenopodium* and *Solanum nigrum* (Marullo 1991; Mouden et al. 2017). During summers, they inhabit the garden plants, like *Liatris* L. cultivars, *Aster novi-belgii* L. and *Aconitum napellus* L. (Mantel and Van de Vrie 1988), feeding on young tissues and causing severe damages to newly developing fruits or flowers.

9.3.2 *Thrips tabaci* (Terebrantia; Thripidae: Thripinae)

Thrips tabaci, commonly known as onion thrips, is another thysanopteran pest species that severely damages the agricultural crops in Asia, Africa, Europe, Australasia and America (Boateng et al. 2014). It is one of those species that exhibit intraspecific variability in terms of wing and body colour, ranging from light yellow to dark brown (Fig. 9.4b). The species has seven antennal segments, and the main vein of the front wing features from four to five distal bristles; however, there are no bristles on the abdominal sternites (Bournier 1983). The adults emerging during winters are darker than the summer counterparts. They reproduce by constant thelitokous parthenogenesis (Dimitrov 1976; Zawirska 1976).

Thrips tabaci emerges from plant tissue through the tunnel drilled by the ovipositor. The females reproduce parthenogenetically and have a number of ecotypes, each being polyphagous and hosted by a wide range of plants. The first instars have fewer antennal segments than the adults. Their integument develops pigmented spots depending on their feeding pattern. Both the first and second larval instars are extremely voracious feeders. The pupal moulting takes place either in soil or on the host plant. *Thrips tabaci* infests more than 300 plant species including cabbage, cucumber, tobacco, vegetables of family Liliaceae, rose, lettuce, potato, tomato, pea, melon and cotton (Gill et al. 2015). It also transmits numerous plant pathogens, like *Iris* yellow spot virus that reduces onion bulb size and quality (Gill et al. 2015).

9.3.3 *Thrips palmi* (Thysanoptera; Thripidae: Thripinae)

Thrips palmi or the ‘palm or melon thrips’ is about 1.3 mm long. It has a pale yellow body with blackish setae (2nd urotergite with four lateral setae, 8th with complete comb in both sexes). The life cycle is slightly different from other thrips species, i.e. second instar larvae move on the ground for development and pupation (Fig. 9.4c). The thrips were collected for the first time from Sumatra (Indonesia) in 1921 from tobacco plants and were introduced to Japan in 1978 (Karny 1925; Sakimura et al. 1986). The potential crops affected by *T. palmi* are cucumber, Chinese spinach, lettuce, watermelon, cantaloupe, sweet pepper, ornamental plants, eggplant, bean species and citrus (Childers and Beshear 1992). It is a polyphagous key pest of solanaceous plants and cucurbits in many temperate and tropical regions. It damages the plants directly by feeding and ovipositing on them or indirectly by transmitting tomato spotted wilt virus (TSWV) (Bragard et al. 2019).

9.3.4 *Thrips fuscipennis* Haliday (Terebrantia; Thripidae: Thripinae)

Thrips fuscipennis is found in Palaearctic region and is generally confused with *T. tabaci*. It differs from the latter in terms of numerous morphological features, including their darker wings and the antennae which are divided into seven segments similar to *T. tabaci*, but the first segment is darker than other segments (Fig. 9.4d).

Three small hairs are seen around the edge of the pronotum in *T. fuscipennis*, but on the back edge of 8th tergite, the hair comb breaks off at the centre. *Thrips fuscipennis* reproduces by thelitokous parthenogenesis. The females overwinter on bark or dry stems but never prefer the soil (Speyer 1932, 1936). They deposit their eggs in sepals and bracts of buds. The numbers of generations developing during reproductive period are not known. Albeit being polyphagous, the pest species prefer roses, but carnations are also frequently infested (Bournier 1983).

9.3.5 *Thrips simplex* Morison and *Thrips meridionalis* Pr. (Terebrantia; Thripidae: Thripinae)

Both the thrips species are morphologically similar. The differences between *T. simplex* (Fig. 9.4e) and *T. meridionalis* (Fig. 9.4f) are that the former has 5–6 distal hairs instead of 3, which are located on the main vein of the front wing, three antennal segments shorter by 20–21 μm and a single row of accessory hairs on 7th sternite instead of two found in *T. meridionalis* (Bournier 1954). Except the base of forewings which are pale brown, the adults of *T. meridionalis* are dark brown or black in colour. Their antennae are eight-segmented, and in the posterior corner of their prothorax, two hairs are found. Further, on their 8th tergite, a comb of hairs is present. Adults of *T. meridionalis* are commonly found on fruit orchard plants (see Tommasini and Maini 1995).

Thrips simplex reproduces by arrhenotokous parthenogenesis (Bournier 1956a). Discovered before 1930 and originating in Australia, this thrips species distributed itself worldwide due to the trade of gladiolus bulbs, which it commonly infests before undergoing overwintering. Females lay their eggs on the bulb, leaves and flowers. *Thrips simplex* begins to reproduce above 12 °C (Bournier 1983) and completes its development within 11 days when reared at 30 °C. Pupation takes place either on the plant, into the soil or amongst the bulb scales. The species may occasionally be found on carnation, freesia, iris, narcissus and tritoma (see Tommasini and Maini 1995).

9.3.6 *Taeniothrips dianthi* Priesner (=*Pesothrips dianthi* (Priesner) (Terebrantia; Thripidae: Thripinae)

Taeniothrips dianthi is morphologically similar to *T. meridionalis*. However, in contrast to *T. meridionalis*, it has a crown of thick and dark hairs around the tip of 3rd segment of its antennae. It is a thermophilic species and was introduced from colder regions of southwest Poland to rest of Europe, where it found compatibility with the environment for its development through carnation trade. After overwintering, the mated females emerge from the soil and settle on carnation seedlings (see Tommasini and Maini 1995). Females lay about three eggs a day. The second larval instars move down the plant to go inside the soil for pupation. During reproductive period, three complete generations are observed (Pelikan 1951).

The adults of *T. meridionalis* are monophagous. They are found only on carnations, causing considerable damage by altering flower morphology and providing a particular appearance called the 'bird's head' (Pelikan 1951; Bournier 1983).

9.3.7 *Heliothrips haemorrhoidalis* (Bouché) (Terebrantia; Thripidae: Panchaetothripinae)

Heliothrips haemorrhoidalis, commonly known as 'Greenhouse thrips', are cosmopolitan and polyphagous insect pests. They have a black body (immature stages are pale) and a red abdominal tip (Fig. 9.5a). The entire body surface is covered by a clearly distinguishable reticular pattern. The legs appear glassy, and the wings form a white contrast against the black body when at rest. Antennae have eight segments. The first two segments are light brown, while other three segments are yellow. The sixth segment is brown at distal portion; however, the last two segments are pale in colour and filiform (see Tommasini and Maini 1995). Females of *H. haemorrhoidalis* reproduce by obligatory thelitokous parthenogenesis, and

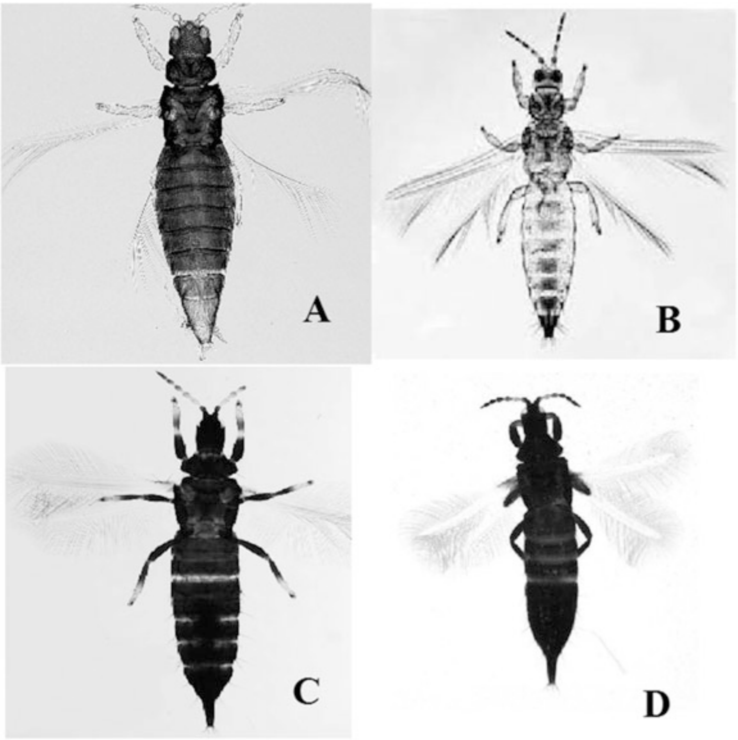

Fig. 9.5 (**a**) *Heliothrips haemorrhoidalis*, (**b**) *Haplothrips tritici*, (**c**) *Liothrips vaneeckei*, (**d**) *Liothrips oleae*

males are extremely rare (Bournier 1956a). Certain females deposit a drop of excrement on their eggs to seal the hole drilled by their ovipositor, while others lay eggs much deeper into the plant tissues. The larval instars secrete a rectal liquid as a part of defence against the potential predators.

Adults overwinter on the bark and fallen dead leaves. Under favourable conditions, the adults feed on many plant species, like *Viburnum L.* spp., *Dracaena* spp., orchids, *Photinia* spp., *Ficus* L. spp., roses, *Croton* L. spp., azalea, avocado, citrus fruit, *Eucalyptus* spp., grapevine and tobacco. In mid-Mediterranean countries, this species is more frequently found on *Viburnum tinus* L. High infestation of *H. haemorrhoidalis* causes complete defoliation of host plant leading to severe economic losses (see Tommasini and Maini 1995).

9.3.8 *Haplothrips cottei* (Vuillet) and *Haplothrips tritici* (Kurdjumov) (Tubulifera; Phlaeothripidae: Phlaeothripinae)

Both *H. cottei* and *H. tritici* are morphologically similar. Adults of *H. tritici* selectively infest the members of Gramineae family (Fig. 9.5b). The females are black and 1.5 mm long, but ends of their front tibia and tarsus are pale. They have hyaline wings that lack veins and are fringed along the margins. The front wings have a double line of 5–8 hairs posteriorly. The 10th segment of the abdomen is long and tube-like. In contrast, the adults of *H. cottei* have longer heads, their 10th abdominal segments (or tube) are shorter and their wings do not exhibit double fringe around the back portion (see Tommasini and Maini 1995). Individuals of *H. cottei* are found in numerous forms, namely, brachypterous at temperatures below 17 °C, macropterous at 30 °C and 87–100% relative humidity and brachypterous at 30 °C and 17% relative humidity (Ghabn 1932). *Haplothrips cottei* particularly infests carnation, is thermophilic in nature and is found in Mediterranean coastal regions, Egypt and Spain.

9.3.9 *Liothrips vaneeckei* Pr. and *Liothrips oleae* Costa (Tubulifera; Phlaeothripidae: Phlaeothripinae)

Both the species are similar in morphology. However, adults of *L. vaneeckei* are black and differ from those of *L. oleae* in colour of their front tibiae which are yellow rather than black. The middle and back tibiae of *L. vaneeckei* (Fig. 9.5c) are yellow at their distal portions. Their abdominal segment is longer, and the third antennal segment is shorter than those of *L. oleae* (Fig. 9.5d). The wings of *L. vaneeckei* are without veins with a double line of 15–18 hairs (see Tommasini and Maini 1995). Both the species are cosmopolitan and reproduce by arrhenotokous parthenogenesis (Bournier 1956b). Bulb-induced contamination of *L. vaneeckei* takes place via windborne intermediaries (Titschack 1960). Adults of *L. vaneeckei* affect lily plants, but the adults of *L. oleae* infest the olive trees.

9.4 Nature of Damage

Majority of the thrips species are highly polyphagous and have an invasive lifestyle (Parker et al. 2013). As a consequence, the adults migrate from one plant to another easily with wind. They are attracted towards yellow, white and blue colours and are easily carried on human skin and clothes as a vector to new agricultural fields (Morse and Hoddle 2006). Both *F. occidentalis* and *T. tabaci* prefer a variety of plant families. Owing to their invasive behaviour, *F. occidentalis* has become cosmopolitan in merely 50 years, since its first description in 1960s in western United States (Reitz 2009; Gill et al. 2015).

Thrips rupture the plant tissue to expose the mesophyll cells, excrete out the enzymes to digest the broken tissues and finally suck the digested compounds. They pierce the plant cells with their piercing- and sucking-type mouthparts and feed on plant juices, imbibing the exuded cellular contents (Koschier et al. 2002). Their feeding creates silver leafing, reduced photosynthesis and environment for pathogens to develop on injured leaves (Reitz 2009; Gill et al. 2015). Studies have shown that *T. tabaci* causes water and chlorophyll loss, provides stress to host plants and injures leaf and bulb tissues leading to reduced bulb size in garlic and onions (Gill et al. 2015). *Frankliniella occidentalis* feeds on young floral bud's tissues and causes large cosmetic damages in cut flowers and sunflowers. Their constant feeding results in development of defected kernels in sunflowers (Chyzik et al. 1995). Besides feeding, the cosmetic damages to crops by *F. occidentalis* are also caused by their oviposition, which leads to holes in plant tissues and growth of pathogens (Reitz 2009).

The most devastating damages caused by thrips are the viruses they transmit to the host plants. *Tomato spotted wilt virus* (TSWV) transmitted by *F. occidentalis* and *Iris yellow spot virus* (IYSV) transmitted by *T. tabaci* are the common plant viruses that severely affect the crop productivity (Mumford et al. 1996). Only 20 plant viruses have been identified so far that are vectored by 14 thrips species (Riley et al. 2011a, b). Tospoviruses are a small class of Bunyaviridae that are spread by thrips to host plants (Mumford et al. 1996). The viruses are generally acquired by the newly hatched thrips feeding on infested plants. Although the late larval instar and adult thrips may acquire the viruses, they cannot transmit it independently (Ullman et al. 1995). The virus moves through the gut of newly hatched larvae/first instars into their salivary glands from where the virus is injected directly into healthy host plants. As an outcome, the plants infected with tospoviruses exhibit the following symptoms: (i) irregular necrotic spots on infected leaves, (ii) black/purple stem streaks, (iii) chlorosis or yellowing of leaves, (iv) chlorotic blotching or chlorotic and necrotic ring spots and line patterns on leaves and fruit, (v) leaf deformation and distortion, (vi) shedding of buds and dropping of leaves, (vii) dieback and collapsing of leaves and (vi) plant death due to wilting (Steenbergen et al. 2018) (Fig. 9.6). Since thrips are poor fliers, they disperse mainly by wind to reach new host plants. They respond to colours, particularly yellow, white or blue colours; UV reflectance; and scent. Their cryptic feeding behaviour and their tendency to burrow deep inside

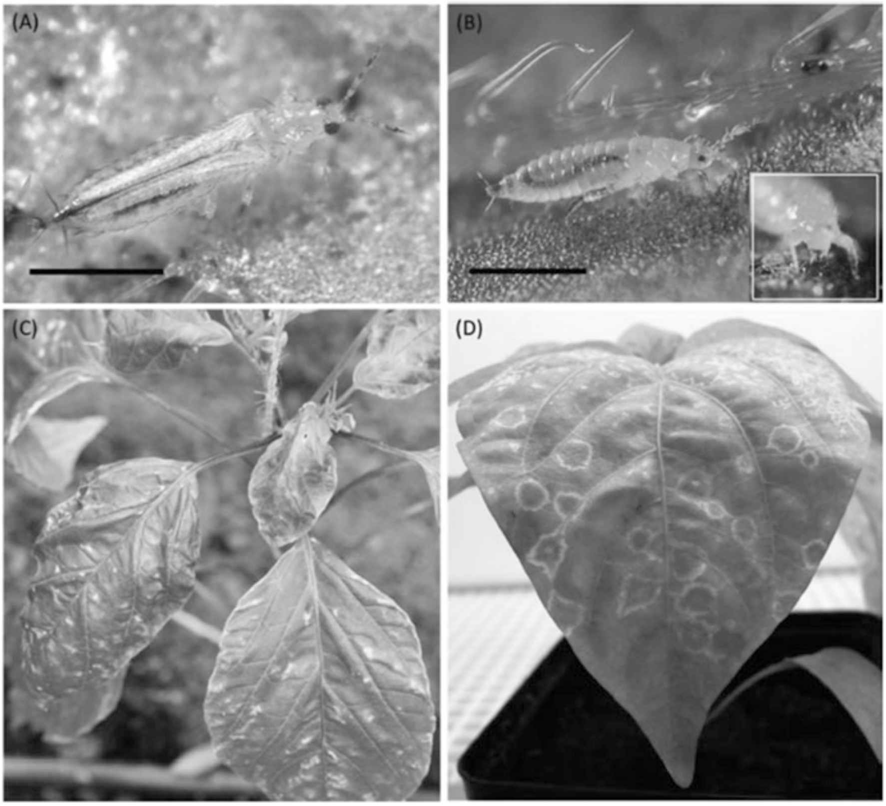

Fig. 9.6 Development of *Frankliniella occidentalis* on pepper *Capsicum* plant (**a**, **b**) and the damage symptoms (**c**, **d**). (see Steenbergen et al. 2018)

plant tissues and hide within them make it difficult to manage them with classical ways of insecticide sprays (Steenbergen et al. 2018).

9.5 Management

9.5.1 Cultural, Mechanical and Physical Control Methods

Farmers in many countries still rely either on cultural or physical methods for the management of thrips. Sanitary practices, viz. removing weeds and old plant materials or debris, form the first line of defence against thrips (Northfield et al. 2008; Nyasani et al. 2013). Studies have shown that the combinations of positive-pressure force ventilation systems with insect-proof screens protect the thrips invasion (Sugiyama et al. 2014). The UV-reflective mulch also repels thrips by interrupting their host-finding behaviour (Reitz et al. 2003; Kigathi and Poehling 2012). While irrigation creates a less favourable environment for thrips, the high relative humidity favours their development (Steiner et al. 2011). Use of fertilizers also increases the abundance of thrips. Studies have shown that increased use of nitrogen and phosphorous fertilizers and high levels of aromatic amino acids increases the populations of *F. occidentalis* (Chow et al. 2012; Chen et al. 2014). Planting trap crops may draw the thrips away from target crops; for example, growing chrysanthemum as trap plant lowers the damages caused by *F. occidentalis* (Cook et al. 2006; Buitenhuis et al. 2007). Similarly, intercropping French beans with sunflower, baby corn or potato reduces the damages to bean pods caused by thrips species (Nyasani et al. 2012).

9.5.2 Host Plant Resistance

Host plant resistance involves complex interactions mediated by morphological and chemical traits of the plant that influence the amount of damages caused by the pest species. Understanding the nature of plant defensive traits may play important roles in designing crop varieties for enhancing their protection against the pests.

Morphologically Defensive Structures The surfaces of host plants act as physical barriers through their morphological traits, like waxy cuticles and epidermal structures, such as trichomes. Studies have further shown that the damages caused by *F. occidentalis* were inversely proportional to the amount of epicuticular wax on gladiolus leaves (Zeier and Wright 1995). The induction of type VI glandular trichomes in response to methyl jasmonate application trapped higher numbers of *F. occidentalis* individuals (Broughton et al. 2015).

Host Plant Resistance Through Defensive Chemicals Defensive plant chemicals arise from both the primary and secondary metabolites. The primary metabolites of host plants or their nutritional chemicals are beneficial for thrips. Amongst different crops, the low concentrations of aromatic amino acids reduce the damages caused by *F. occidentalis* (Mollema and Cole 1996). While studying different chrysanthemum varieties, isobutylamide was associated with *F. occidentalis* host plant resistance (Tsao et al. 2005). The common compounds that constitute resistance ability in plants against the thrips species are jacobine, jaconine and kaempferol glucoside in *Jacobaea vulgaris*; acyl sugars in tomato; chlorogenic and feruloylquinic acids in chrysanthemum; and sinapic acid, luteolin and β-alanine in carrot (Tsao et al. 2005; Leiss et al. 2009, 2013).

Using Transgenic Plants The plant protease inhibitors (PIs) are naturally occurring defensive compounds in host plants that reduce the availability of amino acids for growth and development of insect pests. Transgenic alfalfa, which expresses an anti-elastase protease inhibitor, delays *F. occidentalis* damage (Thomas et al. 1994). Incorporation of purified cystatin and equistatin into artificial diets reduces oviposition rates in thrips (Annadana et al. 2002). Similarly, the expression of multidomain protease inhibitors in potato significantly improves thrips resistance (Outchkourov et al. 2004). Moreover, the use of transgenic plants either modified or simultaneously used with additional management strategies is a promising approach for thrips management. However, the highly limited political and regulatory frameworks restrict the commercialization of genetically modified crops the world over (Mouden et al. 2017).

Inducible Resistance Apart from the constitutive defences, plants also use their inducible defences against pest attacks. The plant hormones, like jasmonic acid (JA), salicylic acid and ethylene, trigger the naturally occurring chemical responses and protect the host plants from insect pests. Studies have shown that JA-responsive genes, i.e. *VSP2* and *PDF1.2*, are strongly stimulated when *Arabidopsis* plants are exposed to thrips (De Vos et al. 2005). Further, treating host plants with exogenous elicitors activates their natural defensive response and enhances their resistance to thrips. Applications of JA on tomato plant result in reduced density, preference and performance of thrips on the host plant (Thaler 1999). Similarly, treating tomatoes with acibenzolar-*S*-methyl (ASM), a functional analogue of salicylic acid, reduces TSWV incidence, but does not influence the population densities of *F. occidentalis* (Pappu et al. 2000). Thus, nowadays, the induced resistance has gained wide publicity and would be of much value in conjunction with other IPM methods.

9.5.3 Behavioural Control

Another strategy in applied pest control is to manipulate the behaviour of insect pests by using semiochemicals, which function as signal compounds. Nowadays, agriculturalists are employing semiochemicals as lures for monitoring and controlling the pests of order Thysanoptera.

Pheromones Neryl (*S*)-2-methylbutanoate is a sexual aggregation pheromone that attracts both males and females of *F. occidentalis* (Hamilton et al. 2005). Decyl and dodecyl acetate are produced as alarm pheromones in anal larval droplets of thrips. The synthetic equivalents of these pheromones cause *F. occidentalis* to reduce their oviposition and landing rates, thereby suggesting their functions as alarm pheromones (MacDonald et al. 2002, 2003). In addition, 7-methyltricosane, a male-specific cuticular hydrocarbon, has recently been identified in inhibiting matings in *F. occidentalis* (Olaniran et al. 2013).

Allelochemicals Various volatile chemicals, like benzenoids, monoterpenes, phenylpropanoids, pyridines and sesquiterpene, attract adult females of *F. occidentalis* in a dose-dependent manner. While thrips are attracted by pure linalool and the linalool emitted by engineered chrysanthemum plants, they are deterred by linalool glycosides (Koschier et al. 2000). Methyl isonicotinate is an attractant for numerous thrips species and is used to locate the host plant (Teulon et al. 2011). Recently, an active ingredient known as volatile (*S*)-verbenone, identified as a lure for thrips, has been described from pine pollen (Teulon et al.

2014). Applications of methyl-jasmonate and cis-jasmone deter the thrips larvae from feeding and settling on host plants (Egger and Koschier 2014; Egger et al. 2014). The monoterpenoid phenols thymol and carvacrol exhibit both the feeding and oviposition deterrent effects (Peneder and Koschier 2011; Allsopp et al. 2014). Combination of dodecyl acetate with maldison, an organophosphorous insecticide, increases the larval mortality of *F. occidentalis* (Cook et al. 2002). Thus, semiochemicals are highly promising mass trapping agents for thrips and may be further used in ‘lure and kill’ methods (Sampson and Kirk 2013; Broughton et al. 2015).

9.5.4 Chemical Control

Insecticides belonging to classes such as organophosphates, carbamates and pyrethroids are generally used to control the thrips. Recently, avermectins and spinosyns isolated from natural soil-borne fungi are the leading insecticide classes to manage the thrips worldwide (Ishaaya et al. 2002; Lebedev et al. 2013) (Table 9.4). Some narrow-spectrum insecticides, like pyridalyl and lufenuron, are also used against thrips. However, frequent applications of broad- and narrow-spectrum insecticides are making the thrips resistant to most of the chemical classes (Bielza 2008; Gao et al. 2012). Henceforth, insecticides may only be used if economic damage thresholds are reached, and their applications should be accurate and precise so that they may not affect the natural enemies (Bielza 2008; Srivastava et al. 2014; Kivett et al. 2015). Moreover, the newly introduced insecticides must be wisely included in thrips management programmes and should be alternated with other insecticide classes to prevent the development of chemical resistance.

Table 9.4 Synthetic and natural compounds used to control thrips

Origin	Type of compound	Target	Crops
Natural	Pyrethrins	Sodium channel	Lettuce, strawberry, cut flowers
	Azadirachtin	Ecdysone receptor	Rose, chrysanthemum, cut flowers
Synthetic	Pyridalyl	Protein synthesis	Rose
	Lufenuron	Chitin biosynthesis	Rose, cut flowers
	Spinosad	Nicotinic acetylcholine receptor	Capsicum, rose, lettuce
	Abamectin	Glutamate-gated chloride channel	Capsicum, rose, chrysanthemum
	Thiamethoxam	Nicotinic acetylcholine receptor	Rose, chrysanthemum
	Methiocarb	Acetylcholinesterase	Rose, chrysanthemum, cut flowers
	Deltamethrin	Sodium channel	Capsicum, lettuce, rose
	Spirotetramat	Acetyl CoA carboxylase	Chrysanthemum

See Mouden et al. (2017)

9.5.5 Biological Control

Biocontrol methods utilize augmentative release of natural enemies and conservation approaches to sustain their efficacy and abundance. Natural enemies of thrips are categorized into two groups, viz. macrobials (parasitoids and predators) and microbials (nematodes and entomopathogenic fungi) (Mouden et al. 2017).

(a) *Macrobials*

In nature, the populations of thrips are regulated by mites and other predatory species belonging to orders Hemiptera, Coleoptera and Neuroptera (Dyadechko 1977). The arthropod predators associated with biocontrol of thrips are phytoseiid mites (*Amblyseius* spp.) and pirate bugs (*Orius* spp.). Predatory mites used against *F. occidentalis* are *Amblyseius barkeri* and *Neoseiulus* (formerly *Amblyseius*) *cucumeris*, which primarily feed upon the first instars of *F. occidentalis*. Species such as *A. limonicus*, *A. swirskii*, *A. degenerans* and *A. montdorensis* are the other effective predatory mites of *F. occidentalis* (Knapp et al. 2013; Buitenhuis et al. 2015).

Predatory anthocorids *Geocoris ochropterus* (Sureshkumar and Ananthakrishnan 1985), *Montandoniola moraguesi* (Varadarasan and Ananthakrishnan 1981) and several species of *Orius* (Vishwanathan and Ananthakrishnan 1974) feed on *Sericothrips variabilis* in soybean fields, while *Carayonocoris indicus* (Sureshkumar and Ananthakrishnan 1984) prey on *Haplothrips ganglbaueri*, *F. occidentalis* and *F. schultzei*. Amongst the different soil-dwelling predatory mites used against thrips, the commercial biocontrol agents are *Stratiolaelaps scimitus* (=*Hypoaspis miles*), *Macrocheles robustulus*, *Gaeolaelaps aculeifer* and the rove beetle *Dalotia coriaria* (=*Atheta coriaria*) (Messelink and Holstein 2008; Wu et al. 2014).

Chalcids, such as *Tetrastichus* and *Pedobius* spp.; eulophids including *Goetheana*, *Thripoctenoides*, *Thripobius* and *Ceranisus*; mymarids, such as *Camptoptera pulla*; and trichogrammatids, such as *Megaphragma* spp., parasitize the thrips (Ananthakrishnan 1984). *Ceranisus americensis* and *C. menes* are the common parasitoid wasps employed to control the thrips species (Loomans 2006). They parasitize the first instar larvae and induce mortality in prepupal stages.

(b) *Microbials*

These include the entomopathogens used against thrips and consist of nematodes and fungi. Nematode species of genera *Steinernema* and *Heterorhabditis* are generally used against soil-inhabiting thrips (Ebssa et al. 2004, 2006). Treatment with *Thripinema* nematodes has shown to induce sterility in females of *F. occidentalis* (Arthurs and Heinz 2006). The conidia of entomopathogenic fungi affect the thrips by penetrating through their cuticle and obtain nutrients for growth and reproduction. Foliar applications of different fungal species, viz. *Metarhizium anisopliae*, *Beauveria bassiana* and *Lecanicillium lecanii* (=*Verticillium*), have resulted in reduced thrips populations in vegetable and floral crops (Ugine et al. 2007). The use of *B. bassiana* has shown sublethal effects on the larvae of *F. occidentalis* (Zhang et al. 2015). Although several formulations of entomopathogenic fungi are available for foliar applications, their efficacies are non-uniform as they require different abiotic conditions (Mouden et al. 2017).

The combinational treatments of different natural enemies or entomopathogens are other alternative treatments for thrips management. Combined applications of *A. swirskii* with *N. cucumeris* in laboratory studies have shown negative interactions in controlling *F. occidentalis* larvae (Buitenhuis et al. 2010). Although combined use of predatory mites and pirate bugs has significantly affected *F. occidentalis* populations, the effects were poor than when a single predator was used (Weintraub et al. 2011). In contrast, a combination of *O. laevigatus* along with a generalist predator, *Macrolophus pygmaeus*, achieved enhanced control of thrips in sweet pepper fields (Messelink and Janssen 2014). *Heterorhabditis* nematodes when used along with foliar-dwelling mite, *N. cucumeris*, provided the best control of thrips in green bean fields (Ebssa et al. 2006). The combinations of different predatory mites with nematodes, like *S. feltiae*, achieved better control of *F. occidentalis* in cyclamen, while combinations of *O. laevigatus* with *S. feltiae* failed to control the thrips (Pozzebon et al. 2015).

9.6 Future Prospects

(i) *Development of Formulations to Target Soil-Dwelling Stage of Thrips*

To date, the microbial control of thrips is largely targeted on adults and larvae inhabiting the crop canopy. However, microbial control approaches are not always successful as majority of the thrips leave their host plants at later stages of their development prior to adult emergence (Hulshof et al. 2003; Berndt et al. 2004). Thus, efforts need to be made in such a direction so as to interrupt the life cycle of thrips by controlling their developmental stages. Moreover, the relatively high humidity of soil offers a more suitable environment to the fungal spores and improves their germination and proliferation for thrips biocontrol (Skinner et al. 2012; Lee et al. 2017). Hence, fungal-based granular formulations may be an alternative strategy that needs to be developed in future.

(ii) *Evaluating Compatibility of Fungal Pathogens with Other Biocontrol Agents*

Recently, numerous fungal pathogens have been developed worldwide for controlling different species of thrips. However, owing to their broad host range, the arthropod pathogens more frequently exert a negative effect on natural enemies and eventually disrupt the pest biocontrol programmes (Thungrabeab and Tongma 2007; Ansari et al. 2008; Ong and Vandermeer 2015). Roy and Pell (2000) reported that certain species of fungal pathogens severely affected the predators when infected hosts were less suitable prey for them. Seiedy et al. (2015) also found that certain strains of *B. bassiana* displayed high pathogenicity to predators of thrips when the hosts were less preferred one. Thus, assessing the compatibility of potential pathogens and beneficial arthropods is highly essential for successful thrips biocontrol. In addition, complementary studies need to be conducted in greenhouse and field conditions to validate the results of laboratory-based bioassay experiments (Mouden et al. 2017).

(iii) *Improving Resistance of Useful Fungal Strains*

Majority of the fungal formulations used against thrips are available for foliar applications. The fungal conidia sprayed on leaves are generally dried out within a short time period or are often inactivated by UV light and high temperature (Rangel et al. 2008; Wu et al. 2013). Therefore, highly resistant fungal strains targeting thrips need to be screened for their wider commercial applications. In addition, the suitable measures to improve fungal resistance are (a) optimizing concentrations of conidial heat-protective saccharides for improving efficacy and germination rates of *B. bassiana* conidia, (b) modifying plant oil as medium for increasing fungal conidial thermotolerance and (c) releasing higher concentrations of glucose as substrates for enhancing conidial thermotolerance and field persistence (Ying and Feng 2006; Kim et al. 2010). Moreover, additional biotechnological tools, like genetic manipulations, should be explored on fungal pathogens to increase their environmental tolerance capacity. In a previous study, Leng et al. (2011) reported that by genetically engineering trehalose pathway, the thermal tolerance capacity of fungal strains was increased without compromising their virulence. Such future studies on similar lines may offer new methods for enhancing thermal tolerance capacity of fungal pathogens.

(iv) *'Omics' Technologies*

The integration of 'omic'-based methods along with the developments in computational systems may provide powerful tools for crop protection. Assessing the interactions between host plants and insect herbivores and understanding genetic variations amongst insect populations and resistant crop varieties may generate useful information for providing new opportunities and technologies for complex resistance traits (Mouden et al. 2017).

(a) *Plant Genomics*

Although the domestication of wild plants through selection has improved yield and palatability, it has greatly reduced the genetic and phenotypic diversity resulting in loss of thrips resistance (Mirnezhad et al. 2009; Leiss et al. 2011). Identifying genes or metabolites as biological markers may help in introduction of novel insect resistance traits into the breeding lines. Presently, successful breeding of resistant cultivars is restricted to control tomato spotted wild virus (TSWV). And the genes responsible for inducing resistance against TSWV are *Sw-5*, *Sw-7* and *Tsw* (Boiteux and De Avilla 1994; Riley et al. 2011a, b).

(b) *Insect Genomics*

The availability of thrips genome data will open up new opportunities in the future to explicate their gene functioning and develop alternative control methods based on molecular interactions of thrips with host plants and/or viruses (Rotenberg et al. 2015). Recently, modulating the functioning of vacuolar ATP synthase subunit B gene has resulted in increased *F. occidentalis* mortality and its reduced fecundity (Whitten et al. 2016). Sequencing salivary gland transcriptome of TSWV-infected and non-infected thrips species may further lead to the assumed explanation of genes involved in detoxification and inhibition of plant defence responses (Stafford-Banks et al. 2014). The accessibility to thrips genome and transcriptome sequence data would further facilitate the development of approaches to either suppress the thrips populations or enhance the plant defensive responses (Mouden et al. 2017).

(c) *Metabolomics*

To date, the use of metabolomics was restricted to comparative approaches using genotypes with contrasting levels of resistance (viz. resistant or susceptible) (Leiss et al. 2011). However, when combined with genetic approaches, the metabolomics results provide useful opportunities to identify the metabolic markers for resistance to thrips and signal the possibilities of 'metabolite breeding'. Thus, identifying compounds providing resistance to different herbivores may form a basis for the multi-resistance breeding programmes. Studies have shown that by manipulating environmental factors, the concentrations of resistance-related metabolites within plants increase, which would ultimately suppress the thrips growth (Demkura et al. 2010). Rutin and chlorogenic acid are two phenolic compounds that are utilized in thrips resistance when plants get exposed to UV-B radiations (Demkura et al. 2010).

Further, the plant secondary metabolites included in thrips resistance may be exploited in future to develop new protective substances that may enhance the defence mechanisms of host plants and minimize the ill effects of chemicals on nontarget species. Beside plants, the microbials may offer huge sources of metabolites for insect resistance. The assemblage of microbial communities may affect the activity of thrips through plant chemistry or volatile emission, because the genetic variations in response of plants to these beneficial bacteria may open the ways for breeding of those plant types that derive the bacterial benefits maximally. However, the above-ground defensive strategies of soil microbial communities on plants against the thrips are yet to be explored (Mouden et al. 2017).

(d) *High-Throughput Screening Methods*

The genomic and/or metabolomics techniques for thrips resistance require a high-throughput screening system. Recently, high-throughput phenotyping methods have been developed that utilize automated video tracking of the behaviour of thrips species (Thoen et al. 2016). However, the reproducible high-throughput methods evaluating the thrips damage are still absent. Similarly, high-throughput screening systems testing for the active plants or microbial metabolites against various thrips species are altogether absent. The stable thrips-derived cell lines have still not been developed beyond the primary cell cultures (Mouden et al. 2017). However, the availability of thrips genome sequence may provide unique opportunity to detect the olfactory or gustatory receptors for developing the basis of high-throughput screening.

9.7 Conclusion

In brief, the integrations of various management strategies are required to control the thrips populations, but such integrations may present significant challenges. Nowadays, biological control methods are gaining wide publicity for managing thrips in protected crops. Another promising strategy is to utilize insect semiochemicals for controlling thrips. In addition, there are numerous biotechnological advances including the 'omics' technologies that would undoubtedly enhance the management of thrips. Still a lot has to be generated about the interactions between host plants and the thrips species, so as to make crucial contributions in developing environment-friendly and biologically sustainable strategies for crop protection. There is a need to accelerate the introduction of new chemicals as biorational insecticides and to enhance the use of natural resources for thrips management. Applying molecular modifications, using genetic engineering tools and developing novel biological products (including microorganisms and metabolites) may probably lead to production of improved cultivars that would possibly respond to thrips attack with enhanced resistance. Further, the existing strategies should be viewed in terms of integrated pest management programmes with economic, ecological and environmental outcomes.

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Mites

10

Abhishek Shukla

Contents

Abstract

Mites are the smallest organism that belongs to phylum Arthropoda, class Arachnida, subclass Acari and order Acarina. Mites occur worldwide and affect human life directly or indirectly. Acari are rich in diversity. Mites occupy every habitat of life. Plant-feeding mites or phytophagous mites attack almost all cultivated crops, ornamental crops and wild plants, thus reducing the yield of cultivated crops drastically and, besides this, serving as vectors for several plant diseases. In agriculture, the problem of phytophagous mites became intense after the introduction and large-scale use of pesticides during the last few decades. This pesticide effectively controlled the insect pests against which they were used but

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A. Shukla (✉)
Department of Entomology, N.M. College of Agriculture, Navsari Agricultural University, Navsari, Gujarat, India
e-mail: abhishekshukla@nau.in

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also destroyed the natural enemies of phytophagous mites. In the absence of the insects for competition for food and other necessities and coupled with elimination of natural enemies such as parasites and predators, the phytophagous mite populations increased to enormous proportions and posed greater problems in addition to the development of resistance and resurgence problems as well as toxic residues. Further, mite species which were never seen earlier have become important pests (e.g. coconut eriophyid mite).

Keywords

Mites · Ecology · Life cycle · Seasonality

10.1 Introduction

Mites, like insects, are omnipresent. They occupy every type of habitats like terrestrial and aquatic. They are found in/on the soil, plants/plant parts, animals and insect. As an arthropod, mites share many special characters with insects, like the presence of an exoskeleton, small size, high reproductive rate, varied reproducing methods and wide adaptability. In recent years, due to indiscriminate usage of pesticides and adoption of several improved agricultural practices, many species of mites which were less important have assumed the status of important creature. Among them, phytophagous mites (plant-feeding mite) and predatory mites prefer to live on plants. The phytophagous mites are responsible for causing severe damage to most of the cultivated crops therein (i.e. chlorophyll losses; stunted growth; stippling of plant parts; webbing on plant leaves, flowers and pods; yellowing of leaves; defoliation; burning of leaves; reduction in fruit size; adverse effect on fruit quality; various types of deformities which appeared on plants; and finally adverse effect on the yield). Moreover, mites indirectly cause adverse effects including decreased photosynthesis and transpiration rate. During daytime, mites spread to all the plant parts when the population increases. However, information pertaining to ecology of mites, especially plant-associated mite, is very scanty. Therefore, the information has been compiled to fill up the lacuna and to have imminent knowledge on the origin and diversity of mites and their interrelation with biotic and abiotic environments wherein they tend to live, reproduce and complete successful growth and development in various ecosystems. Due to the economic losses caused by mite pests, management tactics need to be established to keep population levels under the economic threshold of infestation. This practice should be based on integrated pest management (IPM) including spraying chemical products, using biological control agents and/or resistant varieties, etc.

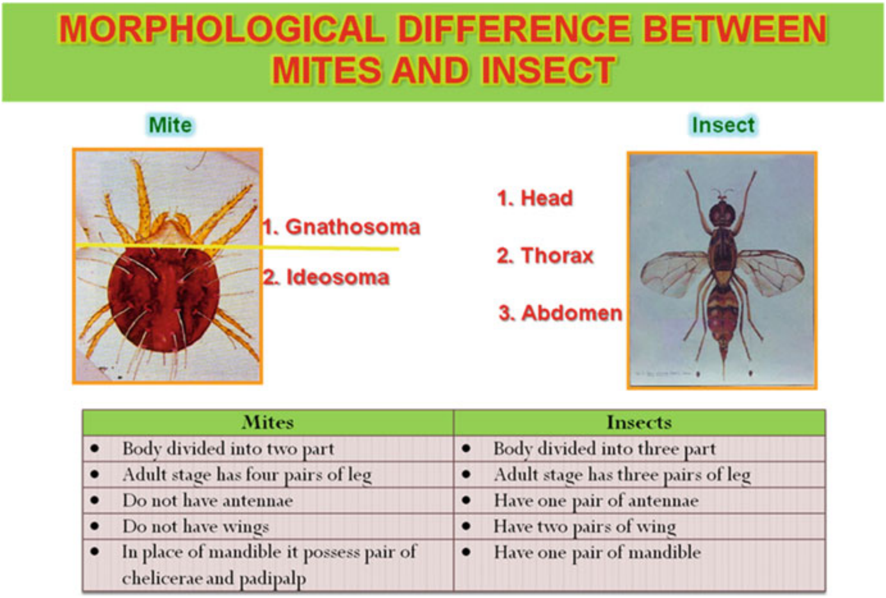
10.2 Origin of Mites

Mites are arachnids and are grouped under Chelicerata along with Xiphosurida (horseshoe crabs) and Eurypterida (marine scorpions). The latter two groups are entirely marine and are almost extinct (except for just four extant species of horseshoe crabs) (Walter and Proctor 1999). Arachnida includes mites along with spiders and scorpions that are terrestrial (with a few secondarily aquatic species). Mites originated between 3.80 and 400 million years ago (mya) probably from Trigonotarbids, the earliest pulmonate arachnids (Jeram et al. 1990). The earliest terrestrial colonist mites were acariform who invade land directly from the ocean and diversified during early Devonian period as major soil macrofauna. Initially, they were microbivores and scavengers and later on spread in their associations with plants and animals (Selden 1993).

10.3 General Characteristics of Plant-Associated Mites

Mites differ significantly from insects (Plate 10.1). Plant-associated mites have specific characteristics, which are outlined below:

1. They are small creatures measuring about 0.1 mm in length.
2. The body is divided into gnathosoma and *Idiosoma*.



MORPHOLOGICAL DIFFERENCE BETWEEN MITES AND INSECT

Mites	Insects
• Body divided into two part	• Body divided into three part
• Adult stage has four pairs of leg	• Adult stage has three pairs of leg
• Do not have antennae	• Have one pair of antennae
• Do not have wings	• Have two pairs of wing
• In place of mandible it possess pair of chelicerae and padipalp	• Have one pair of mandible

Plate 10.1 Morphological difference between insects and mites

3. The word gnathosoma is derived from Greek word *gnathos* = 'jaw' and *soma* = 'body', one of the important parts of the body of Acari (mites and ticks), and it comprises the mouth and feeding parts. The feeding parts include the hypostome, the chelicerae and the pedipalps. The gnathosoma is separated from the main body of the mite, i.e. the *Idiosoma*, by circumcapitular furrow or circumcapitular suture which is a flexible section of the cuticle.
4. Two pairs of mouthparts such as chelicerae and pedipalps are adapted to grasping, piercing, sucking and tearing the host/prey.
5. Legs are two segmented with chelate or pedipalps.
6. Larval stage possesses three pairs of legs, whereas four pairs of legs are found in adults (exception: eriophyid mite possesses two pairs of legs in its adult stage).
7. The respiration takes place by tracheas which are connected with stigmata. In some cases, it may be cutaneous.
8. Metamorphosis is simple.
9. Normally, the male is smaller in size than female.

10.4 Important Species of Plant-Associated Mites

10.4.1 Phytophagous Mites

Among all arachnids, majority of them are carnivorous or detritivorous (spiders, scorpions, ticks and mites), except a few that are phytophagous in nature (less than 10%). In comparison, there are nearly 40% of phytophagous insects (Mitter et al. 1988). There are hardly 5000 species of phytophagous mites compared to over 4, 00,000 species of phytophagous insects. One reason for this small percentage of phytophagy among mites might be due to low morphological diversity with respect to their mouthparts. The phytophagous mites belong to the order Acariformes under Prostigmata generally feeds on foliage of vascular plants suck plant fluids by using their stylet-like mouthparts. Lindquist (1996) believed that phytophagy organisms probably evolved from mycophagy organisms. Eriophyid mites: Most of the described species of phytophagous mites belong to one family, Eriophyidae (2900 species), and the remaining to two other families of Eriophyoidea: Diptilomiopidae and Phytoptidae. The mouthparts of eriophyid mites consist of 7 to 9 stylets encased in a 'U'-shaped channel of the subcapitulum but do not form a feeding tube. Two methods of attachment to the leaf surface during feeding are observed. Most eriophyoids have the apices of the palp tarsi modified into a suction pad that allows them to anchor their mouthparts into the leaf epidermis. The muscles of the palps then contract and force the stylets into the epidermal cell of the host (Nuzzaci and Alberti 1996). In contrast, some mites anchor themselves to the leaf surface with anal suckers, arch the body and force the stylets into epidermal cells (Oldfield and Michalska 1996). Formation of galls, leaf deformation, petioles, stems, buds, flowers, fruits and appearance of blisters are very common symptoms that appear

Plate 10.2 *Tetranychus urticae* infestation on brinjal

with infestation by eriophyid mites. The most striking symptoms in some cases is erineum which are appearance of felt-like pelts of hair like processes letteron they usually turn into reddish in colour. Mites mostly live among the leaf hairs and appear to feed on them. The exact mechanism causing the galling or appearance of erineum is not well understood but appears to result from the release of some chemical that the mite induces during feeding (Nuzzaci and Alberti 1996). The cell-punctured tetranychoid mite dies, but the surrounding cells begin to proliferate to form a protective gall: Spider mites and false spider mites belong to Tetranychoidea that contains five families of phytophagous species. Some species in the subfamily Tetranychinae are producing silken webbing, and due to this they are commonly known as spider mites. Various *Tetranychus* species produces the largest and most complex web and often in dense entanglement of several layers. Some species, like the two-spotted spider mite, *Tetranychus urticae*, are polyphagous with over 150 hosts (Belavadi 2010; Plate 10.2, 10.3 and 10.4).

The family Tetranychidae is the largest (1200 species) and is the most important group, damaging several cultivated crops. The Tenuipalpidae (false spider mites) with over 600 species in 22 genera (Evans et al. 1993) is another important group of phytophagous mites. The remaining three families, Tuckerellidae, Linotetranidae and Allochaetpphoridae, account for less than 30 species and are agriculturally least important (Belavadi 2010; Shukla et al. 2013). Unlike the other groups of plant-feeding prostigmatid mites, the Tetranychoidea appears to have evolved from predaceous ancestor like Cheyletoidea or Raphignathoidea (Krantz 1978; Evans 1992). No cheyletid mite feeds on plants, but a number of predatory cheyletids are associated with plants, and probably some descendants of these mites started feeding on plant cells. The mites mainly causing economic damage to food crops are spider mites (family: Tetranychidae), false spider mites (family: Tenuipalpidae), gall mites (family: Eriophyidae), yellow or broad mites (family: Tarsonemidae) and peacock mites (family: Tuckerellidae). The eriophyid mites are to a great extent host specific

Plate 10.3 *T. urticae* infestation on okra

Plate 10.4 Spider mite attacked marigold

among all the phytophagous mites, while the mites of other families mentioned below are not host specific. Some of the important features of these groups of Acari are as follows:

10.4.1.1 Spider Mites (Family: Tetranychidae)

Spider mites are soft-bodied, medium-sized mites and measured about 0.5 to 0.6 mm, with various colour morphs, and they feed on the undersurface of the leaves by forming colony as they are exclusively plant feeders. Many species spin webs like spiders to cover their colonies, hence named as spider mite, except *Bryobia* and *Petrobia*. When they are alive, they are often red, green, orange or yellow in colour.

Table 10.1 List of phytophagous mite families in India with the number of species and genera under those along with the number of species on agricultural crops and the number of pests

Family/ superfamily	Number of species	Number of genera	Number of species on agriculture crops	Total number of pests	Total number of major pests	Total number of minor pests
Tetranychidae	106	20	75	19	8	11
Tenuipalpidae	93	20	40	6	1	5
Eriophyidae and	500 (+)	100 (+)	20	13	6	7
Rhyncaphytoptidae						
Tarsonemidae	10	5	5	3	1	2
Tuckerellidae	4	1	2	1	–	1
Total	**718 (+)**	**146 (+)**	**142 (+)**	**42**	**16**	**26**

Table 10.2 Losses caused by plant mites (Anonymous 2009)

Crops	Losses	Crops	Losses
Mango	50–80%	Vegetables	10–15%
Litchi	30–40%	Tea	5–11%
Paddy	20–25%	Ber	25–30%
Sugarcane	20–30%	Rose	25–30%
Red gram	15–30%	Carnation	25–30%
Cotton	20–30%	Gerbera	25–30%

The gnathosoma has a capsule-like structure known as the stylophore, which is formed by the fusion of chelicerae. The movable digit of the chelicerae is very long, often whip-like and recurved proximally. A pair of stigmata is located near the base of the chelicerae, where the peritremes arise. The palps are five-segmented. The spinneret size and shape is of great taxonomic significance. The *Idiosoma* is often covered with a striate cuticle. The pattern of the striation and the shape/density of lobes distributed on the striae are useful diagnostic characters. Wedge-shaped males are smaller than ovoid females and have a tapering opisthosoma. Males have a protrudable aedeagus; the shape is very important in species identification. Larval stage possesses three pairs of legs, while nymph and adult stages possess four pairs of legs. This family is represented by two subfamilies: Bryobinae and Tetranychinae. Spider mites occur on almost all of the major agri-horticultural crops as well as infest most of the ornamental plants in almost all environments where mainly Tetranychinae species can potentially cause economic damage (Tables 10.1 and 10.2).

10.4.1.2 False Spider Mites (Family: Tenuipalpidae)

They are quite similar to spider mites, but they are unable to spin webs. These mites are pear shaped and flat and measured about 0.3–0.35 mm; they are mostly bright coloured and move slow and are normally found on the undersurface of leaves, twigs, fruits, etc. Some false spider mites live inside the galls and feed on barks also. They possess four pairs of legs. Because most of the species are dorsoventrally flattened, the false spider mites are also known as flat mites. They are usually found on the lower surface of the leaves near the midrib or veins, and they are slow-moving mites. Some species feed on the bark, while others live under leaf sheaths or in galls or in flower heads. Only a small number of species belonging to a few genera have become pests of economic plants, and they are most commonly found on tropical fruit crops and ornamental plants. The false spider mites are mostly found in tropical and subtropical parts of the world, and it includes about 800 well-described species which belongs to over 25 genera. This family consists of three subfamilies: Tegopalpinae, Brevipalpinae and Tenuipalpinae; the two latter subfamilies, i.e. Brevipalpinae and Tenuipalpinae, include most of the described species. The two largest genera *Brevipalpus* and *Tenuipalpus* are considered as the most economically important pests of agri-horticultural crops.

10.4.1.3 Yellow or Broad Mites (Family: Tarsonemidae)

They are yellow or glossy in colour and tiny, fast-moving mites which measured about 0.25–0.30 mm in size. They are found on undersurface of leaves as well as many times associated with fungus. Female deutonymphs are often carried by males. The adults possess four pairs of legs. The family Tarsonemidae is widely distributed and possesses about 545 species of 45 genera. The family Tarsonemidae is represented by three subfamilies, which are Pseudotarsonemoidinae, Acarapinae and Tarsoneminae. The subfamily Tarsoneminae includes most of the described species which comes under the two large genera, i.e. *Tarsonemus* (over 270 species) and *Steneotarsonemus* (over 70 species). Tarsonemid mites exhibit various feeding habits; some species feed on fungus, algae and plants; and some of them can prey on parasite insects. Some plant-feeding tarsonemid mites are pests of agri-horticultural crops, most of them belonging to a few genera in the Tarsoneminae, except for the *Polyphagotarsonemus.* Since these mites feed on surface cells, more significant damage is observed in young tissues of the host plant. Leaf discoloration with a silver aspect is characteristic symptoms produced by this mite. The mite-infested developing leaves became shriveled or curled and eventually shed, and severely attacked plants stop growing. Occasionally, plant tissue ontogeny is also altered due to toxins injected during the process of feeding. Worldwide, the chilli mite or yellow mite or broad mite or jute mite, *Polyphagotarsonemus latus* (Banks), is certainly the most important pest of many crops and ornamental plants in field or greenhouses. The dispersal of this mite species take place by wind, by human transport of infested plant parts, and most importantly through insects living on plants (phoratic manner by whiteflies) in chilli and capsicum. Tarsonemid mites that belong to the genus *Steneotarsonemus* are phytophagous and specialised on monocotyledon plants.

10.4.1.4 Peacock Mite (Family: Tuckerellidae)

The sapota fruit mite, *Tuckerella kumaonensis* Gupta, was recorded on sapota fruits causing significant qualitative damage. Previously, it was considered being a minor pest of sapota. However, there were complaints from sapota cultivators that the surface of the fruits becomes blackish which cannot be washed with water and such fruits are considered to be inferior quality. This blackening on fruit was due to the attack of the mite, *T. kumaonensis*. Under the south Gujarat agroclimatic conditions, the sapota fruit mite remains active round the year with peak activities during the month of April (Shukla et al. 2013). The attack of sapota fruit mite was more severe on Kalipatti and Cricket Ball varieties (Shukla and Radadia 2015a, b). The red-coloured adult mite sucks the cell sap from the fruit surface starts from marble size fruit stage, results in to the fruit surface rough. The affected fruit surface becomes rough or black or dark coloured which results in qualitative loss of harvested products. The family Tuckerellidae is one of five families within the superfamily Tetranychoidea that also includes the spider mites (Tetranychidae), false spider mites (Tenuipalpidae), Allochaetophoridae and Linotetranidae. The mites belonging to these five families are obligately phytophagous. The mouthpart structures and feeding habit of tuckerellid mites appear to be quite similar to other tetranychids.

10.4.1.5 Gall Mites (Family: Eriophyidae)

They are tiny, measured about 0.20–0.25 mm in size, elongated, and worm-like with only two pairs of legs. Apart from leaves, they also live within buds and may form gall, and hence they are also called as gall mite. The superfamily Eriophyoidea is distributed worldwide. From all over the world, about 3000 species belonging to over 250 genera are known. The superfamily consists of three families: Phytoptidae, Diptilomiopidae and Eriophyidae. About three-quarters of the described species in Eriophyoidea belong to the Eriophyidae. Eriophyid mites are tiny worm-like or fusiform mites, and they form galls or live freely on various host plants. The wounding and injecting of specific salivary secretions into host cells result in a specific response of the affected leaf, stem or bud tissues, such as gall differentiation, hypersensitive reaction or non-distortive feeding effects and in some cases complex symptoms, considered as syndromes. Most species are monophagous, and many species are limited to plant species within a single genus with few exceptions. Most species cause little harm to their host plants; however, some rust mites and gall mites are important pests on economic plants.

10.4.2 Beneficial Mites (Predatory Mites)

Beneficial mites have significant importance in achieving biological control of phytophagous mites. Predatory mites mainly belong to the families Phytoseiidae, Stigmaeidae, Cunaxidae, Bdellidae, Cheyletidae, Erythraeidae, Tydeidae, etc. Among them, the family Phytoseiidae plays important role in regulating phytophagous, while the rest are of relatively less importance. Important genera of the beneficial mites are *Phytoseiulus*, *Galendromus*, *Mesoseiulus*, *Amblyseius*, *Neoseiulus*, etc. Among them, phytoseiid mites are predatory in habit and feed on phytophagous mites and small insects, so they are very useful in biological control of phytophagous mites and small insects on crops (Chant and McMurtry 2007). Phytoseiid species occurs from tropical rain forests to arctic tundra, and they have been recorded from all significant land masses, except from Antarctica (Chant and McMurtry 2007). They inhabit a wide range of terrestrial habitats like plants, leaf litter, logs, fungal colonies, mosses, bird nests, mammal burrows, stored products and soil (Gowda and Gupta 2010). The phytoseiid mites have extensively exploited the foliage habitat of higher plants among mesostigmatids (Chant and McMurtry 2007). On plants, they normally inhabit the undersurface of the leaves, especially near the veins. They also inhabit flowers, fruits and area under the bark. They show a wide range of food habits, feeding on animal to nonanimal foods such as pollen, honeydew, nectar, etc. These mites feed on crawlers of scale insects, eggs of certain species of moths, whiteflies, thrips, aphids, etc. Under unavailability of prey, these mites feed on plant nectar and honeydew from aphids, soft scales, whiteflies, mealy bugs, etc. Various kinds of fungi may also serve as food. Because of the wide range of food habits, these mites are better adapted to survive when their preferred food is not available. Hence, these mites are more effective as well as efficient as predators (Gowda and Gupta 2010).

10.5 Life History

Mallik et al. (2004) recorded that female coconut eriophyid mite, *Aceria guerreronis* Keifer (Eriophyidae), laid white and oval-shaped eggs. The nymphs are small and resemble the adults in shape. The total developmental period is 7–8 days. The mites continue to reproduce and multiply underneath the perianth. Further, the biology of sapota fruit mite, *T. kumaonensis* Gupta, on sapota fruit was studied under laboratory conditions at Navsari, South Gujarat, India. The study revealed that both the sexes had four developmental stages (larva, protonymph, deutonymph and tritonymph) interrupted in between with a quiescent stage. The average incubation period was 11.12 ± 1.74 days, which were followed by larval period of 10.75 ± 1.48 days. The nymphal stages passed through three instars, viz. protonymph, deutonymph and tritonymph, with mean duration of 10.30 ± 1.34, 10.85 ± 1.95 and 10.80 ± 1.50 days, respectively. In adult stage, the pre-oviposition, oviposition and post-oviposition period varied from 11.95 ± 1.23, 22.60 ± 4.10 and 12.05 ± 2.52 days, respectively. On an average, the adult period was 40.75 ± 2.937 days, and a single female in its entire life span laid on an average 19.35 ± 4.44 eggs. During developmental period, the maximum mortality was noticed in larval stage (17.58%), and it was followed by egg stage (9.00%). The fully grown adults were measured 0.347 ± 0.006 mm in length and 0.171 ± 0.008 mm in width, respectively (Shukla and Radadia 2015a, b). Moreover, the biology of two-spotted spider mite, *Tetranychus urticae* (Koch), was studied on gerbera cv Stanza in three seasons, i.e. summer, monsoon and winter, by Shah and Shukla (2014). The developmental duration from egg to adult stage for males was 18.08 ± 2.47, 21.54 ± 1.60 and 25.36 ± 1.59 days, while for unmated females it was 28.54 ± 2.87, 30.18 ± 2.55 and 53.65 ± 4.85 days; further, for mated females, it was 24.81 ± 2.57, 23.88 ± 1.66 and 52.15 ± 5.04 days during three seasons, respectively. On an average, unmated female laid 26.06 ± 8.33, 31.86 ± 6.01 and 19.13 ± 3.85 eggs during her life span, whereas on an average a mated female laid 59.24 ± 13.84, 71.80 ± 15.45 and 66.48 ± 10.72 eggs in her life span during three seasons, respectively. The average fecundity was 25.77 eggs per unmated female and 64.50 per mated female. On an average, the time taken for completion of the life cycle of the mite on gerbera leaves was 21.66 days for male, 37.47 days for unmated female and 37.29 days for mated female. The biology of *T. urticae* on tomato was studied by Pokle et al. (2016) in three seasons, i.e. summer, monsoon and winter. The findings revealed that on an average the life cycle of the spider mite on tomato leaves completed in 30.34 days for male, 39.85 days for unmated females and 44.00 days for mated females. A single unmated female laid 30.76 eggs in her life span, whereas a mated female in her life span laid 87.38 eggs. Sex ratio of bisexual reproduce was 1:2.48 with 71.32% females and in case of parthenogenetically reproduces only males were produced (100% males). Patel et al. (2016) studied the biological attributes of *T. urticae* on marigold in winter season under south Gujarat agroclimatic situation. The life cycle of *T. urticae* passed through five different stages, viz. egg, larvae, protonymph, deutonymph and adult stage, as well as three quiescent stages, viz. nymphochrysalis, deutochrysalis and teliochrysalis. Egg incubation period was 4.76 ± 1.48 days. The larval, protonymph, deutonymph period and adult period were 2.84 ± 0.80, 3.56 ± 1.19, 3.04 ± 0.84 and 9.88 ± 1.45 days for male and 5.36 ± 1.11, 4.52 ± 1.11, 4.04 ± 0.93 and 18.44 ± 1.92 days for female, respectively. The male-to-female ratio of bisexual reproduction was 1:2.43, whereas unmated female produces 100% males. Pre-oviposition, oviposition and post-oviposition period lasted for 4.72 ± 1.06, 7.12 ± 1.17 and 6.04 ± 0.84 days for mated female and 5.72 ± 1.43, 5.64 ± 1.11 and 4.12 ± 0.83 days for unmated female, respectively. Mated and unmated females on an average produce 22.68 ± 5.13 and 64.68 ± 9.63 eggs. The total life period of *T. urticae* was shorter in male (21.96 ± 2.26 days) compared to mated female (40.55 ± 3.12 days). The length of eggs, larva, protonymph and deutonymph were 0.12 ± 0.01, 0.14 ± 0.01, 0.20 ± 0.02 and 0.29 ± 0.03 mm and the breadth were 0.12 ± 0.01, 0.09 ± 0.01, 0.12 ± 0.01 and 0.14 ± 0.02 mm, respectively. The length and breadth were 0.42 ± 0.03 and 0.18 ± 0.01 for male and 0.47 ± 0.02 and 0.22 ± 0.02 for female, respectively.

10.6 Seasonality in Plant Mites

According to Shukla et al. (2013), the sapota fruit mite, *Tuckerella kumaoensis* Gupta, population gradually increased during 14th SMW (5.60 mites/fruit) and reached to its peak in 18th SMW (7.30 mites/fruit), but after that the fruit mite population showed a gradual declining trend. In mid-August season (32nd to 34th SMW), the mite population disappeared. The correlation studies of the 3-year pooled data suggested that the fruit mite population showed a significant positive correlation with maximum temperature, whereas it has a negative correlation with minimum temperature, morning RH and evening RH. In a study under polyhouse conditions on gerbera, Shah and Shukla (2014) reported that the two-spotted red spider mite, *T. urticae*, remained active round the year and caused damage to gerbera. The maximum spider mite population was noticed during last week of July 2013

(12.86 mites per leaf). The correlation studies between weather parameter and spider mite population showed that the spider mite population had a nonsignificant negative correlation with minimum, maximum and average temperature, while spider mite population exhibited a significant positive correlation with average relative humidity. On polyhouse gerbera, the spider mite population was maximum on top strata as compared to middle and bottom strata. Further, Pokle and Shukla (2014) noticed that the population of russet mite, *Aceria lycopersici*, remains active throughout the year under polyhouse conditions with peak activities of egg and mobile stages during 21st SMW (fourth Week of May). Further, a significant positive correlation existed between both eggs and mobile stages of russet mite with average temperature and average relative humidity under polyhouse conditions on tomato. The maximum number of eggs and mobile stages was seen on top leaves canopy of the crop. Later on, Shukla and Radadia (2015a, b) conducted field experiment during *Kharif* 2013 and 2014 at Navsari to investigate the population dynamics of rice leaf mite *Oligonychus oryzae* on two rice varieties, i.e. Jaya and Gurjari. The activities of leaf mite were first noticed in 21st SMW (last week of May) and were reached to its peak during the month of August on both rice varieties. However, it was also noticed that the leaf mite disappeared at the time of harvesting of the crop. Kakde et al. (2016) carried out field experiment to study the population dynamics of sheath mite, *Steneotarsonemus spinki* Smiley, infesting rice under two types of rice cultivation systems, i.e. conventional planting and SRI method. The results pertaining to the conventional planting method showed that mite first appeared during the first week of September (35th SMW) with 0.80 mites/leaf sheath. It gradually increased and reached peak (18.04/leaf sheath) during the first week of October (40th SMW). It declined in the second week of October but was recorded till harvest (43rd SMW). In case of SRI planting method, the mite incidence started from first week of September (35th SMW). Further, it gradually increased and reached peak (11.95/leaf sheath) during the first week of October (40th SMW) and recorded till harvest (43rd SMW). Thus, mite incidence started from booting stage, increased gradually and reached peak during dough and milky stage. The mite population was lower in SRI method as compared to the conventional method. The pooled data indicated that the maximum temperature and bright sunshine exhibit positive effects on the mite population, while minimum temperature and relative humidity had negative effects on the mite population. Additionally, the seasonal activities of two-spotted red spider mite, *T. urticae*, infesting polyhouse French bean were studied by Chauhan and Shukla (2016) at Navsari, Gujarat, during January 2015 to June 2015. The study revealed that the spider mite population starts to build up during the first week of January (first SMW) (4.33 mites/leaf); later on, the spider mite population gradually fluctuated and reached to the first peak during third week of February (eighth SMW). Further, the spider mite population again fluctuates during different time intervals, and a second peak was clearly noticed during the first week of May (19th SMW). The spider mite population showed a nonsignificant negative correlation with average temperature, whereas the same spider mite population exhibited a nonsignificant positive correlation with average relative humidity under polyhouse condition. In polyhouse, throughout the crop season, the distribution of *T. urticae* was higher on top strata, and it was followed by middle and bottom strata.

Mahendrakumar and Shukla (2016a) conducted a field experiment during January to June 2015 to investigate the population dynamics of spider mite, *T. urticae*, on brinjal (cv Pant bahar). The study revealed that the population of spider mite, *T. urticae*, remains active throughout the crop season. The spider mite population reached to peak during the last week of April (18th SMW). A significant positive correlation exists between spider mite population and maximum, minimum and average temperature, whereas a significant positive correlation exists between spider mite population and its predators, viz. predatory mite and ladybird beetle. The study revealed that the spider mite, *T. urticae*, preferred middle leaf canopy of brinjal plant. In a study, Desai et al. (2017) investigated the seasonal incidence of spider mite *T. urticae* on rose under polyhouse condition under South Gujarat agroclimatic condition during 2015–2016. The data on the seasonal incidence of *T. urticae* revealed that the spider mite population was observed from the third week of July to the first week of August. The spider mite population exhibited highly significant positive correlation with minimum temperature and average temperature, while it exhibits a nonsignificant negative correlation with average relative humidity and maximum temperature.

10.7 Economically Important Mite Species

Agriculturally important mite species can exhibit either phytophagous or predatory habits. Most important taxa of Acari exclusively include phytophagous mites, which are Eriophyoidea and Tetranychoidea (Tetranychidae, Tenuipalpidae). Tarsonemidae is an important family which includes several species of mite pests. All these Acari taxa include the most important species of mite crop pest which are distributed worldwide and several mite species which are geographically restricted. The superfamily Eriophyoidea is very largely distributed worldwide. These fusiform or worm-like mites cause various forms of plant abnormalities such as galls, leaf blisters and rusts. Most eriophyid mite species are monophagous or are limited to plant species within a single genus. Some rust mites and gall mites are important pests on economic plants. The Tetranychidae which is also known as spider mites is a large family which includes approximately 1200 species belonging to over 70 genera and is distributed worldwide. Spider mites cause mechanical damage by sucking cell content from leaves. At first, it shows up as a stippling of light dots on the leaves, and sometimes leaves became bronze in colour. As feeding continues, the leaves turn yellow and drop off. Often leaves, twigs and fruit are covered with a large amount of webbing. Tenuipalpidae, which is also known as false spider mites or flat mites, has approximately 800 species and is described in over 25 genera. Only *Raoiella, Brevipalpus* and *Tenuipalpus* and few other genera become pests of economic plants, mainly on tropical fruit crops and ornamental plants. Some mite species belonging to Eriophyidae or Tenuipalpidae has been documented as vector of different plant virus diseases like pigeon pea sterility mosaic disease.

These mites cause economic losses in crops; due to the economic losses caused by mite pests, management tactics need to be established to keep population levels under the economic threshold of infestation. This practice should be focused on integrated pest management (IPM). Since the evolution of pesticide resistance in phytophagous mites is very common, therefore, in current scenario, chemical control has to deal with serious control failures in mite populations. Consequently, different chemical molecules are being currently developed to face this phenomenon, mainly in spider mites. However, the low level of immigration of susceptible individuals and the rapid reproductive rate associated to these mite groups have made it difficult to manage population in crops.

Biological control is an environmentally safe, cost-effective and energy-efficient pest control, either on its own or as a component of integrated pest management. Although several mite species belonging to Bdellidae, Cheyletidae, Cunaxidae, Stigmaeidae and Tydeidae have shown predatory habits, Phytoseiidae mites have been more widely included in biological control programmes, due to their capacity for surviving and reproducing on other arthropods. Additionally, some phytoseiid mites have shown to be resistant or less susceptible to chemical compounds commonly used to control pest mites in commercial crops, thus making them suitable for their use in integrated pest management programmes.

10.8 Reasons for Mite Pest Outbreak

(i) Random and indiscriminate use of chlorinated hydrocarbons which eliminate natural enemies of pest mites, leading to rapid multiplication of pests.
(ii) Development of pest resistance due to uses of sublethal doses of organophosphoric compounds, carbamates, etc., and often due to resurgence problem caused by injudicious use of synthetic pesticides especially pyrethroids.
(iii) Use of high-yielding crop varieties, adapting improved agricultural practices and excess irrigation which make the crops more nutritive and that lead to more mite multiplication.
(iv) Insufficient coverage of pesticides with the habitat of plant-feeding mites.
(v) Many of the mites which were earlier innocuous or of little importance have assumed the status of major pests now throughout the world.

10.9 Nature of Damage

10.9.1 Direct Damage

Mites penetrate the leaf tissues through their needle-like chelicerae and feed on the exudate fluid. While doing so, they damage leaf tissues, destroy chlorophyll (causing reduction in photosynthesis rate), cause rapid loss of water, etc. Plants often remain covered with dense webs where dust particles adhere to, and this affects normal physiological activities of plants. All these produce typical mite damage symptoms as follows: appearance of light yellow spots which develop into greyish leaf colour changes ‘from yellow to brown’, often curling and crinkling appear, leaves become brittle, trees suffer premature defoliation, plants wither, growth gets checked and quality and quantity of fruits and flower formations are affected. Normally, these symptoms are seen in spider mites and false spider mites.

Some eriophyids and tenuipalpids inject toxins through saliva while feeding which causes malformations and deformations (citrus, guava), gall formations (pongamia), erineum (brinjal, litchi), witch’s broom effect (mango, chrysanthemum), blisters (sugarcane), russetting (citrus), etc.

10.9.2 Indirect Damage

Some mites act as vectors of plant viral diseases like pigeon pea sterility mosaic disease (by *Aceria cajani*), fig mosaic disease (by *Aceria ficus*), wheat streak mosaic disease (by *Aceria tulipae*), dolichos enation mosaic virus (by *Tetranychus ludeni*) and sugarcane streak mosaic virus (*Aceria sacchari*).

Some mites are also known for causing fungal diseases.

10.10 Management Measures

10.10.1 Cultural Control

It has long been known that high humidity levels reduce the reproductive potential of tetranychids whose optimal environment is provided by hot and dry air (Sabelis 1986; Duso et al. 2004). A report in 1969 demonstrated that host plant nutrition may have a positive effect on the reduction of the mite population by varying the fertiliser regime applied to the crops (Markkula and Tiittanen 1969). Furthermore, large quantities of nitrogen or a deficiency of potassium can increase the amount of soluble nitrogen available in the plant so that sharp increases in the populations of *T. urticae* follow such fertiliser regimes (Sabelis 1986). However, the plant response to such extreme feeding regimes is not economically viable, so variations in host nutrition have not been used for pest control (Helle and Sabelis 1985).

10.10.1.1 Host Plant Resistance

Research to find sources of resistance to *mites* has been carried out on a variety of crops like soybean, cucumber, *Vigna sp.*, strawberry (Miller et al. 1985), watermelon, maize, tomato, French bean, brinjal, rose, gerbera, carnation, marigold and citrus (Shoorooei et al. 2013). Several studies have found differences in susceptibility to the mite between different cultivars or selections. However, the resistance may be polygenic in most cases (Easterbrook and Simpson 1998) and so is difficult to exploit by plant breeders. Even partial resistance is potentially useful in IPM programmes, however, as it slows the rate of population increase of the spider mite and so makes it easier for predators to gain control. Mechanisms of host plant resistance to *mites* have been attributed to flavonoid pathways in citrus (Agut et al. 2014), leaf trichomes on *Fragaria* (Olbricht et al. 2014), increased peroxidase and polyphenol oxidase activity in melon, antibiosis and antixenosis in bean (Kamelmanesh et al. 2010) and phytochemical compounds in watermelon. El-Saiedy et al. (2011) reported a negative relationship between mite infestation and tannins and nitrogen and protein content in maize leaves (Mead et al. 2010).

10.10.2 Biological Control

Some important natural enemies of phytophagous mites are as follow:

10.10.2.1 *Phytoseiulus persimilis* (Athias-Henriot) (Acari: Phytoseiidae)

Biological control of spider mites using predators is commonly practised, most often using Phytoseiidae (Ferrero et al. 2011). *P. persimilis* is a specialist predator that feeds exclusively on tetranychid mites and whose survival depends on the presence and quality of its prey. This mite is very voracious, with adults able to eat 34 eggs or 4.8 females and 10 juveniles of *T. urticae* per day (McMurtry and Croft 1997). *P. persimilis* can colonise 10 plants in 24 h, with only the ground and the connections between the plants influencing the movements of this predator (Helle and Sabelis 1985). However, there are some failures in the biological control of *T. urticae* by this predatory mite. *P. persimilis* is sensitive not only to the quality and quantity of prey but also to environmental conditions, such as temperature and relative humidity. A relative humidity of 90% is necessary for all eggs to hatch, with the percentage of hatching approaching 0 at 50% relative humidity. Temperatures between 30°C and 35 °C result in optimal development (Sabelis 1986). In tomato crops, *P. persimilis* is hampered by molecules released by the glandular trichomes, such as triacylglycerols and some sesquiterpenes, and its effectiveness in this culture is not satisfactory (Kennedy 2003). The successful control of *T. urticae* with *P. persimilis* depends largely on maintaining the correct balance between predators and prey (Helle and Sabelis 1985).

10.10.2.2 *Neoseiulus californicus* (McGregor) (Acari: Phytoseiidae)

The use of predator like *N. californicus* has also been investigated. This polyphagous predator feeds on the *T. urticae* along with other mite species (Castiglioni et al. 2002). It can survive for a few days without eating any prey by feeding solely on a diet of pollen. In addition, its life cycle is not hampered by low relative humidity, but it is less voracious than *P. persimilis* and suffers from a low birth rate. However, augmentative or inoculative use is strongly considered, and this mite is currently marketed by several firms (Rondon et al. 2005).

10.10.2.3 *Neoseiulus longispinosus* (Evans) (Acari: Phytoseiidae)

This is the most widely studied species in India. The mean of immature period for egg, larvae, protonymph and deutonymph were 2.010 ± 0.341, 0.851 ± 0.129, 1.152 ± 0.354 and 2.666 ± 0.515 days, respectively, when fed on *T. urticae*. The total developmental period completes in 6.240 ± 0.890 days, while the pre-oviposition, oviposition and post-oviposition period took 1.601 ± 0.287, 18.600 ± 2.610 and 3.500 ± 1.007 days. The period of adult longevity for male and female was 9.760 ± 1.116 and 21.080 ± 2.856 days, respectively. A single female laid on an average 38.040 ± 4.631 eggs in her life span when reared on *T. urticae* (Mandape et al. 2018a). The feeding potential showed that larva, protonymph, deutonymph and adult male and female of the predatory mite, *A. longispinosus*, consumed 13.27 ± 1.32, 11.34 ± 1.23, 10.87 ± 1.02, 12.60 ± 2.32 and 15.74 ± 1.45 eggs of *T. urticae*; 5.30 ± 0.64, 5.74 ± 0.57, 6.80 ± 0.84, 5.60 ± 1.07 and 13.70 ± 0.84 mixed stages of *T. urticae*; and 2.44 ± 0.52, 3.84 ± 0.61, 4.34 ± 0.49, 5.44 ± 0.49 and 9.34 ± 0.81 adults of *T. urticae*, respectively. During the entire life period, the male predatory mite consumed 48.07 ± 3.48, 23.43 ± 1.34 and 16.03 ± 1.09 eggs, mixed stages and adults, respectively, while the female predatory mite consumed 56.27 ± 3.37, 31.54 ± 1.16 and 19.93 ± 1.75 eggs, mixed stages and adults of *T. urticae*, respectively. The larva, protonymph, deutonymph and adult male and female of *A. longispinosus* showed more preference towards eggs followed by mixed stages and adult stages of prey mite (Mandape et al. 2018a, b).

10.10.2.4 *Amblyseius alstoniae* Gupta (Acari: Phytoseiidae)

The mean of immature period for egg, larvae, protonymph and deutonymph were 2.010 ± 0.341, 0.851 ± 0.129, 1.152 ± 0.354 and 2.666 ± 0.515 days, respectively, when fed on *T. urticae*. The total developmental period completes in 6.240 ± 0.890 days, while the pre-oviposition, oviposition and post-oviposition period took 1.601 ± 0.287, 18.600 ± 2.610 and 3.500 ± 1.007 days. The period of adult longevity for male and female was 9.760 ± 1.116 and 21.080 ± 2.856 days, respectively. A single female laid on an average 38.040 ± 4.631 eggs in her life span when reared on *T. urticae* (Mandape et al. 2018a, b).

10.10.2.5 *Feltiella acarisuga* (Vallot) (Diptera: Cecidomyiidae)

Another commercially marketed and interesting predator is *F. acarisuga*, which is widely used in greenhouses. Cecidomyid females lay their eggs within spider mite colonies, and the predaceous larvae eat eggs, nymphs and adults. F. acarisuga can be released in combination with the predaceous mite, *P. persimilis*, but *F. acarisuga* is more mobile in the adult stage than *P. persimilis* and eats at least five times more spider mites once it is established. However, *P. persimilis* is known to eat midge eggs if prey is limited, so they should not be released where *F. acarisuga* is already established (Gillespie et al. 1998).

10.10.2.6 *Stethorus punctillum* (Weise) (Coleoptera: Coccinellidae)

The coccinellid, *S. punctillum*, has been reported to be efficient in seeking out spider mite colonies both in open field and greenhouse. Each female beetle is capable of laying about 100 eggs in her lifetime. Both the larvae and adult beetles feed on all stages of the spider mites and their eggs (Rott and Ponsonby 2000).

10.10.2.7 *Stethorus pauperculus* (Weise) (Coleoptera: Coccinellidae)

The biology of coccinellid beetle, *S. pauperculus*, was studied at Navsari, Gujarat. It passes through four stages, viz. egg, larvae, pupae and adults. The incubation period was 4.48 ± 0.35 days when fed on *O. indicus*, while it was 4.20 ± 0.51 days when fed on *T. urticae*. Larvae were found to pass through four instars. The newly hatched grub was dark brown in colour, very tiny and active with a number of long, brown and grey hairs all over the body. The average duration of the first, second, third and fourth instar larvae was 1.55 ± 0.31, 1.28 ± 0.25, 1.83 ± 0.22 and 2.80 ± 0.24 days, respectively, when fed *O. indicus*, while the same was 1.67 ± 0.24, 1.25 ± 0.25, 1.91 ± 0.42 and 2.55 ± 0.41 days, respectively, when fed on *T. urticae*. The average pupal period when fed on *O. indicus* was 3.90 ± 0.20 days and 3.75 ± 0.25 days when fed on *T. urticae*. The average adult period for male was 29.97 ± 2.33 days when fed on *O. indicus*, and it is 27.17 ± 3.10 days on *T. urticae*. The female lives for 32.63 ± 1.83 days when fed on *O. indicus* and 30.63 ± 2.38 days when fed on *T. urticae*. A single female on an average laid 357 ± 55.17 eggs when fed on *O. indicus*, while when fed on *T. urticae* it laid 307.23 ± 21.89 per female. The total life period of male predator was 35.93 ± 4.74 days on *O. indicus*, whereas it is 30.43 ± 3.35 days when fed on *T. urticae*; in female the total life period was 46.23 ± 6.90 days on *O. indicus* and 49.00 ± 4.63 days on *T. urticae* (Godhani and Shukla 2015a). For complete life cycle, the predator *S. pauperculus* required 548.57 ± 11.00 eggs or 38.20 ± 6.60 larva or 275 ± 6.46 nymphs or 150.07 ± 6.88 adults of *O. indicus*, whereas in case of *T. urticae* it required 544.20 ± 8.28 eggs or 324.97 ± 6.46 larvae or 249.07 ± 7.65 nymphs or 132.43 ± 4.36 adults (Godhani and Shukla 2014). The predator population showed two peaks, i.e. the first peak during 40th SMW (fifth to tenth October 2012) with 4.20 predators per plant, whereas the second peak was observed during 17th SMW with 2.73 predators per plant (26th April to second March 2013) on the sorghum. It was also observed that during the peak activities of predators, the population of prey (*O. indicus*) was also highest. The correlation studies between the predator and prey with abiotic factors suggested that there was a significant positive correlation existed between predator *S. pauperculus* and prey *O. indicus* ($r = 0.964$). The maximum, minimum and average temperature had positive significant correlation with the population of predator, whereas morning, evening and average relative humidity had a negative correlation with predator population. The rainfall had negative significant correlation ($r = -0.283$) with the predator population in sorghum under the field conditions. The observation on the population dynamics of *S. pauperculus* on *baru* grass was also recorded because *baru* grass is a very common monocotyledons weed in sorghum as well as found on bunds and is an alternative host of sorghum mite, *O. indicus*. The predator remained active throughout the year on *baru* grass, and it showed two peaks during the year. The first peak was recorded during 43rd SMW (last week of October 2012) (3.33 predators/plant), while the second peak was observed during 17th SMW (last week of April 2013) (4.67 predators/plant). The prey mite, *O. indicus*, was also higher during the period. The correlation between predator *S. pauperculus* and its prey *O. indicus* was also worked out, and a positive correlation ($r = 0.974$) existed between them, whereas correlation between various abiotic factors and predator was also investigated. There was a positive significant correlation between predator and maximum, minimum and average temperature ($r = 0.615$, 0.498 and 0.703), whereas a nonsignificant positive correlation existed between morning, evening and average relative humidity. A negative correlation exists between predator population and rainfall ($r = -0.156$) (Godhani and Shukla 2015b). The pesticides like acephate 75% WP and ethion 50% EC were toxic to the larvae, while the application of azadirachtin 0.15% was least toxic to the larvae of *S. pauperculus* under field conditions (Godhani et al. 2016). In brinjal, the activities start in mid-January and reached to peak in the first week of March, and the predator remained active throughout the crop period (Mahendrakumar and Shukla 2016b).

10.10.2.8 *Scolothrips longicornis* Priesner (Thysanoptera: Thripidae)

Several species of predatory thrips of the genus *Scolothrips* have been investigated for their potential to contribute to the control of spider mites, specifically *S. longicornis* (Pakayari et al. 2011). Pakayari et al. (2009) observed that *S. longicornis* seems to be a good predator against *T. urticae* from late spring to early fall in greenhouses in the Mediterranean region where the temperature is frequently above 25 °C.

10.10.2.9 Microorganisms (Bacteria and Fungi)

Many previous studies have underlined the moderate effect of *Bacillus thuringiensis* against *T. urticae* and its predators (Macintosh et al. 1990, Chapman and Hoy 1991). Aksoy et al. (2008) proved that the biotype B of *Pseudomonas putida* had strong efficacy in causing the mortality of *T. urticae*. Further, Aksoy and Mennan (2004) demonstrated the effects of a fluorescent *Pseudomonas* sp. isolate against nematodes. However, none of the fluorescent *Pseudomonas* have been used as a biocontrol agent of mites to date, although an infection that caused disease in *T. urticae* was identified as *Pseudomonas aeruginosa*. Indeed, the susceptibility of mites to fungal infections could rapidly lead to efficient management of spider mites by some entomopathogenic fungi (Maniania et al. 2008). In such a case, *Neozygites* spp. could be used either via inundative release or by augmentative release. According to Moino et al. (1998), the application of *Beauveria bassiana* Balsamo and *Paecilomyces fumosoroseus* Apopka on *T. urticae* results in high rates of mortality. In addition, these fungi could be used with *P. persimilis* and *N. californicus* as well as garlic extracts as part of an integrated pest management strategy. However, the susceptibility of mites to entomopathogenic fungi varies between different isolates of the fungus (Braga et al. 2009).

10.10.3 Chemical Control

The synthetic acaricides used in the control of *T. urticae* are characterised by a large variety of chemical structures and modes of action found among the many types of compounds that are toxic to phytophagous mites (Pinochet 1991)). The modes of action of acaricides and the identification of their target sites were reviewed by Knowles (1997) and Dekeyser (2005). An overview of acaricides is listed according to their group, their primary site of action and the emergence of resistance based on Van Leeuwen et al. (2009) and the Insecticide Resistance Action Committee (IRAC 2008) (Table 10.3). Although the number of compounds available for the control of mites looks impressive, in practice the number of acaricides that are registered for use in particular crops can be rather limited (Van Leeuwen et al. 2009).

10.11 Conclusion

Mites are microscopic creatures and possess two/four legs in adult stages. However, larval/nymphal stage possesses three pairs of legs. The phytophagous mites possess good diversity with prevalence of the natural enemies. Phytophagous mites mainly causing economic damage to cultivated crops are spider mites (family: Tetranychidae), false spider mites (family: Tenuipalpidae), yellow or broad mites (family: Tarsonemidae), gall mites (family: Eriophyidae) and sapota mites (family: Tuckerellidae). Predatory mites mainly belong to the families Phytoseiidae, Stigmaeidae, Cunaxidae, Bdellidae, Cheyletidae, Erythraeidae, Tydeidae, etc. Among them, family Phytoseiidae plays important role in regulating phytophagous mites, while the rest are of relatively less importance. In modern agro-ecosystems, population of mites reaches extremely high densities if right management interventions are not be applied. Thus, on the basis of the correlation studies, it is very much clear that hot and dry conditions during summer are highly favourable for multiplication of mites. Under stress situation, mite outbreak can be observed on some of the susceptible crops. Rainfall followed by turbulent winds causes major reduction in plant-invading mites. No single factor is responsible in mite population fluctuation though all the factors work in compliment with each other. In most of the cases, the mite population was maximum on top strata as compared to middle and bottom strata. Mites have a short life cycle, and many overlapping generations are developed during favourable season. Nowadays, mites are serious threat to various agro-ecosystems and difficult to manage based on sole management strategies especially acaricide-based management options. Prevailing natural enemies could be best utilised as biological control agents for the management of phytophagous mites by incorporation into the IPM programme to achieve the goal of sustainable agriculture (Tables 10.4, 10.5, 10.6, 10.7, 10.8 10.9, 10.10).

Table 10.3 Mite pests emerging threat agri-horticulture

Family: Tetranychidae	
Red mite, *Tetranychus ludeni* Zacher	It is also a major pest of vegetable in the summer (May to June). It is widespread in the warmer parts of the world and occurs on a great variety of cultivated and wild plants. This mite has been identified as very damaging to cowpea in the central and eastern India. Attacked leaves initially present yellow patches, which eventually become necrotic. Aggregation of mites on tips of the pods is common during periods of the day when temperature is above 38°C. This mite always prefers the lower leaf surface. At high population levels, copious webbing covers the entire plant, leading the mites to migrate
Vegetable mite, *Tetranychus macfarlanei* Baker & Pritchard	It builds up small colonies on the lower leaf surface. Infestations are higher on mature than on young crops
Vegetable mite, *Tetranychus neocaledonicus* André	This mite is a serious pest on brinjal, and it was reported from sponge gourd, bitter gourd, beans, tomato and potato. It was also recorded throughout the year on fig and mango trees (Singh et al. 2000). It builds up small colonies on the lower leaf surface
Sorghum mite, *Oligonychus indicus* (Hirst)	Feeding of this mite causes the appearance of reddish spots on sorghum and sugarcane, which increase with severity of attack. These spots later coalesce to form large red patches, which eventually may cover the whole leaf
Brown wheat mite, *Petrobia latens* (Muller)	This is a serious pest in dry land agriculture, reaching usually higher densities on wheat than on barley. Coriander, cumin and fennel were also found infested by this species. Population levels are highest in March. It is most common under rainwater-fed cultivation. It is mostly common in Rajasthan, Punjab, Haryana, Western Uttar Pradesh and some wheat growing areas of Madhya Pradesh
Oriental mite, *Eutetranychus orientalis* (Klein)	This is a major pest of citrus and sometimes reported from pear, peach, ber, cucurbits, papaya and cotton, damaging the upper surface of fully grown leaves. Population of this mite increases on apple from March to June. Infestations start along the midrib, with mites moving farther away as population size increases. The webbing produced by the mites traps dust particles, giving the upper leaf surface a dirty appearance
Guava mite, *Eotetranychus hirsti* Pritchard & Baker	It is greenish yellow with black blotches all over the dorsum**.** Infested leaves drop prematurely, often resulting in complete plant defoliation. Fruits may also drop. It is an alarming pest of fig in Delhi, Punjab, West Bengal and Uttar Pradesh. This mite is host specific, hence known as fig spider mite

(continued)

Table 10.3 (continued)

Tea and coffee mite, *Oligonychus coffeae* (Nietner)	This is one of the serious pests of tea, coffee and mango in India. It is the most alarming pest of tea in the northeastern region of the country. Banerjee and Cranham (1985) reported that *O. coffeae* reached outbreak level more commonly in northern than in southern India and Sri Lanka. They have two population peaks a year, the first between March and May and the other in September to October. It normally feeds on the upper surface of old leaves, but in case of severe infestation in the summer, the lower surface and the young leaves are also infested. The mites live under the webbing they produce and are found throughout the year
Mango mite, *Oligonychus mangiferus* (Rahman & Sapra)	It infests the upper leaf surface. This is an important pest of mango in different mango growing regions
Apple mite, *Panonychus ulmi* (Koch)	Adults of this species are easily recognised in the field by their brick red colour. This mite has been collected in most of apple orchards of Himachal Pradesh, reducing plant growth and fruit quality. Its infestation is so high that ICAR has established a centre at Mashobra (Shimla) to tackle the problem of *P. ulmi* in the region.
Sugarcane web mite, *Schizotetranychus andropogoni* (Hirst)	This is a very detrimental pest of sugarcane in the northern part of the country. The pest causes the formation of white blotches on the leaf, which gradually dry out. The webbing it produces turns brown and at a later stage it is blown off by wind, dispersing the mites
Family: Tenuipalpidae	
Flat mite, *Brevipalpus californicus* (Banks)	This mite has been collected on citrus and papaya from different states of India. It is the best known species of flat mite and has long been known as a citrus pest. Adult mites feed on the lower leaf surface, mostly along the mid-rib. Feeding causes yellow spots that later coalesce to form brown patches. In case of heavy infestation, the mites congregate at the base of fruits, and their feeding may result in premature fruit drop
Red and black flat mite, *Brevipalpus phoenicis* (Geijskes)	This is a polyphagous mite species. These mites are usually aggregated along the leaf midrib. Infested leaves turn pale yellow and develop brownish patches, dropping off prematurely. Attacked guava fruits show brownish patches. Although extremely rare, males of this species were also collected on guava plants
Pineapple false spider mite, *Dolichotetranychus floridanus* (Banks)	This mite attacks plants of all ages but causes greater damage to young plants. It feeds on the soft white tissue at the base of the leaf. They produce rust-like spots that can be infected by bacteria and fungi, so that rotting of the infected tissues may occur. Severely infested plants may remain small, bearing no fruits

(continued)

Table 10.3 (continued)

Ber gall mite, *Larvacarus transitans* (Ewing)	In Rajasthan State, this mite was collected in blisters of ber plants, *Ziziphus mauritiana*. The twigs of bushy ber cultivars were found heavily infested by this mite. On the average, 20–80% of the bushes were infested. Distinctive characters of this mite are the presence of three pairs of legs in both nymphal and adult males and females. In Rajasthan State, ber bushes were once believed to be free from insect and diseases, but in the survey conducted in 1996, alarming problems caused by this mite were recorded, including the presence of blisters all around tender twigs. The galls rupture during monsoon. After emerging from the galls, the mites hide themselves in cracks and crevices of tree trunk, starting new infestations. The intensity of gall formation was higher on local than on improved cultivars. Twigs bearing galls bore less fruits than healthy twigs
Family: Eriophydiae	
Pigeon pea eriophyid mite, *Aceria cajani* Channabasavanna	The sterility mosaic is a serious disease of pigeon pea, which is an important source of protein in vegetarian diets. The virus causing this disease is vectored by *A. cajani*. This mite was recorded in August and September at low population levels and in March at high levels, when the crop is about to mature. The symptoms include general stunting and yellowing of infected plants, proliferation of branches, leaf mosaic mottle, reduction in leaflet size and partial or complete plant sterility
Coconut eriophyid mite, *Aceria guerreronis* (Keifer)	It causes premature dropping of young nuts, imparting discoloration or scars on their surfaces, taking shelter beneath the floral bracts. The injury begins around the floral bracts and extends down to the inner husks or fibres of coconut. The injured area becomes shriveled brown and scattered. This mite was first noticed in coastal states of South India in 1998. The sudden problem posed by it has jeopardised the cultivation of coconut in Tamil Nadu and Kerala States, in that part of India. The appeal made by coconut growers attracted the attention of Indian Parliament and ICAR
Litchi mite, *Aceria litchii* (Keifer)	Mite causing erineum on the underside of leaves. In the beginning of the attack, the leaves look greener than normal, later turning light to deep brown with velvety hair growth. The deep brown colour indicates that the erineum is drying and that the mites are abandoning it to move to other leaves. The affected leaves become distorted or curled and eventually dry and fall off. This mite has jeopardised the cultivation of litchi (*Litchi chinensis*), a cash crop grown extensively in Muzaffarpur area of Bihar and some parts of Assam State

(continued)

Table 10.3 (continued)

Tomato russet mite, *Aceria lycopersici* (Wolff)	The first sign of damage is the curling of the lower leaves and appearance of a silver gloss on the lower leaf surface; later, infested leaves become bronze coloured, withering and eventually dying. Before damage is noticeable on the top most leaves, their lowermost portion loses the surface hairs. The mites also feed on the stem, whose colour changes from green to brown, cracks appearing on the surface. High levels of this mite are observed on brinjal and tomato in the summer
Mango eriophyid mite, *Aceria mangiferae* Sayed	Both vegetative and floral malformations can occur, the former being the most common on seedlings. However, yield reduction is caused mainly by floral malformations, because of the formation of dense masses of flower buds and the consequent reduced fruit set. Mango malformation is a serious malady of mango in North India. This problem has been related to this mite and to a fungus
Ber woody gall mite, *Eriophyes cernuus* Massee	This mite causes woody galls usually on one side of the stem of jujube plants. In jujube orchards, plants often suffer with heavy and bulky galls distributed over the tree. Galls are small on younger trees but may be large in older trees. The size of the galls may be of 2 to 3 kg. Thousands of mites can be collected by rupturing a small portion of a gall
Family: Tarsonemidae	
Chilli mite, *Polyphagotarsonemus latus* (Banks)	Chilli has emerged as one of the cash crops; the cultivation of this crop in progressively larger scale has coincided with increasing infestation levels of this mite and with increasing crop losses. The damage shows up as bronzed and distorted new growth, with curled leaf margins. Leaf hairs become prominent, the lower surface of the leaves turns reddish and the plant becomes russetted. Starting at the tip, the plant withers and auxiliary buds then produced are killed (Tambera disease). Similar effects are produced on red pepper plants, and early infestation prevents flower and fruit development, while later infestations cause flower drop (Murda disease)
Rice sheath mite, *Steneotarsonemus spinki* Smiley	Infested leaf sheath turns brown, whereas infested panicles produce chaffy or discoloured grains. In recent years, rice sheath mite is gaining a serious pest status in the rice-growing areas of India, especially on fine rice varieties
Family: Tuckerellidae	
Sapota fruit mite, *Tuckerella kumaonensis* Gupta	It is also known as sapota fruit mite and very common in Southern Gujarat. The mite is responsible for qualitative loss of fruits as the fruits become rough giving bronzing appearance

Table 10.4 Life cycle of key phytophagous mites

Crop	Mite pests	Period	Duration (days)			Fecundity
			Egg	Young	Adult	
Cotton	*Tetranychus urticae* Koch	April 1974	3–4	4–10	5–18	2–13
Cotton	*Tetranychus macfarlanei* B & P	1980–1981	3–4	7–12	15–28	50–120
Sorghum	*Oligonychus indicus*	March–August 1986	3–8	3–10	3–14	12–54
		September–October 1996	3–4	5–7	5–10	33–58
Okra	*Tetranychus macfarlanei*	March 1988	4–5	5–7	5–15	10–44
		July 1988	3–4	5–6	10–22	46–100
Brinjal	*Tetranychus urticae* Koch	March 1993	3–5	4–5	9–12	23–32
Chilli	*Polyphagotarsonemus latus* Banks	June 1997	2–3	5–7	4–13	28–39
Indian Bean	*Tetranychus cinnabarinus* Boisd.	January–March 1989	4–7	7–11	3–18	5–50
Indian bean	*Tetranychus ludeni* Zacher	February 1993	4–6	6–9	12–16	38–48
Cowpea	*Tetranychus ludeni* Zacher	May–June 1997	2–4	4–6	5–8	43–66
Little gourd	*Tetranychus macfarlanei* B& P	March 1993	3–5	7–12	4–18	14–64
		July 1993	3–4	8–11	11–14	25–38
		December 1993	4–6	12–15	5–15	20–43
Pigeon pea	*Eotetranychus broodryki* Meyer	March–April 1989	4–5	5–6	3–11	3–20
Coconut	*Oligonychus sp.*	March–April 1989	3–4	4–7	2–7	1–6
Sapota	*Tuckerella kumaonensis* Gupta	March 1994	9–13	58–75	33–55	9–23
Rice	*Steneotarsonemus spinki* Smiley	September 2003	2–4	0.5–2.0	5.0–9.0	15–32
Carnation	*Tetranychus urticae*	September 2008	2.5–5.0	8.49–14.14	11–17	38–106
Rose	*Tetranychus urticae*	August 2008	2–5	7.7–13.5	10–16	38–106
Chrysanthemum	*Tetranychus urticae*	March–April 2010	2–5	7.6–13.5	7–14	35–95
Sorghum	*Oligonychus indicus*	August 2011	2–4	5.4–10.1	11–26	30–60
Mango	*Tetranychus fijiensis*	June 2011	3–6	9.3–13.3	7–15	45–112
Gerbera	*Tetranychus urticae*	June 2012	3.3–4.3	9.8–13.3	9–11.3	7.2–13.7
Tomato	*Tetranychus urticae*	July 2013	2.6–4.6	21.3–26.5	14–18	77.9–96.6

(continued)

Table 10.4 (continued)

Crop	Mite pests	Period	Duration (days)			Fecundity
			Egg	Young	Adult	
Marigold	*Tetranychus urticae*	June 2014				
French bean	*Tetranychus urticae*	August 2015	2–7	10.4–25.4	6–9	39–48
Brinjal	*Tetranychus urticae*	May 2015	3–4.5	7.25–15.9	15–26	46–86
Rice	*Steneotarsonemus spinki* Smiley	August 2016	2–3	3–7	8–18.5	15–33

Table 10.5 Incidence of phytophagous mites on different crops and their peak (s)

Crop	Mite pest	Study period	Period of occurrence	Peak activity
Cotton	*Tetranychus urticae*	1972–1974	Oct–Nov	Oct (third week)
Cotton	*Tetranychus macfarlanei*	1979–1981	Sept–Mar	Dec–Jan
Brinjal	*Tetranychus urticae*	1992–1993	Mar–June	May (second week)
Cowpea	*Tetranychus ludeni*	1992–1993	Apr–July	May (fourth week)
Little gourd	*Tetranychus macfarlanei*	1992–1994	Throughout the year	Dec (second week)
	Brevipalpus californicus			April–May
Sapota	*Tuckerella kumaonensis*	1993–1995	Throughout the year	April (1 fortnight)
Cowpea	*Tetranychus ludeni*	1998	Mar–June	May (second week)
Mango	*Oligonychus mangiferus*	1988–1990	Jan–Sept	Feb
	Cisaberoptus kenyae			Jan–Mar
Rose	*Tetranychus urticae*	1998–2001	Throughout the year	May–June
Okra	*Tetranychus macfarlanei*	2000–2003	Apr–July	June (I fortnight)
Chilli	*Polyphagotarsonemus latus*	2000–2003	Sept–Feb	Oct (I fortnight) Dec–Jan
Sorghum	*Oligonychus indicus*	2000–2003	Sept–Dec	Oct (II fortnight)
Pigeon pea	*Aceria cajani*	2000–2003	Aug–Feb	Oct (II fortnight) Jan (I fortnight)
	Eutetranychus broodryki	2000–2003	Sept–Feb	Dec
Rice	*Steneotarsonemus spinki*	2002–2003	Sept–Oct	October (II fortnight)
Carnation	*Tetranychus urticae*	2008–2009	Feb–Sept	July (first week)
Rose	*Tetranychus urticae*	2008–2009	Feb–Sept	July (first week)
Sapota	*Tuckerella kumaoensis*	2009–2012	Throughout the year	18 SMW
Chrysanthemum	*Tetranychus urticae*	2009–2010	Oct–March	February (last week)
Sorghum	*Oligonychus indicus*	2010–2011	Sept–Dec	October (first week)
Mango	*Tetranychus fijiensis*	2010–2011	Oct–Sept	June (second week)
Gerbera (polyhouse)	*Tetranychus urticae*	2010–2016	Throughout the year	27 SMW and 35 SMW

(continued)

Table 10.5 (continued)

Crop	Mite pest	Study period	Period of occurrence	Peak activity
Orchids (polyhouse)	*Tenuipalpus pacificus*	2010–2016	Throughout the year	September (last week)
Gerbera	*Tetranychus urticae*	2011–2012	Feb–Jan (polyhouse)	July (first week)
Carnation (polyhouse)	*Tetranychus urticae*	2011–2014	Throughout the year	April (first week)
Tomato	*Tetranychus urticae*	2012–2013	Jan–July	April (second week)–June (second week)
Tomato	*Aceria lycopersici*	2012–2013	Jan–July	May (fourth week)
Marigold	*Tetranychus urticae*	2013–2014	April–Jan	July (last week)
Rice	*Oligonychus oryzae*	2013–2014	Throughout crop period	August (first week)
Pigeon pea	*Eutetranychus broodryki*	2013–2015	32–52 SMW	32 SMW
Capsicum (polyhouse)	*Polyphagotarsonemus latus*	2013–2016	41–09 SMW	February (second week)
French bean	*Tetranychus urticae*	2014–2015	Jan–July (field condition)	May (first week)
French bean	*Tetranychus urticae*	2014–2015	Jan–July	April (last week)
Brinjal	*Tetranychus urticae*	2014–2015	Jan–June	May (first week)
Rose (polyhouse)	*Tetranychus urticae*	2015–2016	Throughout the year	29 SMW
Rice	*Steneotarsonemus spinki* Smiley	2015-2016	Aug–Oct	October (third week)

Table 10.6 Varietal susceptibility based on mite population density and damage intensity in different crops under field condition

Crop	Mite pests	Susceptibility			Mode of evaluation
		Less	Moderate	High	
Okra	*Tetranychus macfarlanei*	Padra 18-6, Pun. Padmini, Guj. Bhendi 1, Sel. 2,	Parbhani Kranti, H.B. 55	Red 1 Foot Long, K.S. 312, H.B. 57	Population density
Brinjal	*Tetranychus urticae*	ABH-2, Pusa Purple Long, Ringan Gulabi	PLR-1, M-62, DBR-8, Doli-5	ABH-1, Morvi 4-2, M-36	Population density
Sapota	*Tuckerella kumaonensis*	Co-1, Co-2	Zumakhiya, Cricket Ball	Pillipatti, Mohanguti, Kalipatti	Population density
Mango	*Oligonychus mangiferus*	Vashi Badami, Dadamiyo	Sardar	Totapuri, Alphonso	Population density
	C. kenyae	Vashi Badami	Jamadar, Sardar	Kesar, Alphanso, Vanraj, Dadamiyo, Banarasi Langara, Neelam	
	Aceria mangiferae	Jamadar, Kesar, Totapuri, Vashi Badami	Sardar	Alphonso, Dadamiya, Karanjio	
Pigeon pea	*Sterility mosaic disease (SMD)*	ICPL 87119, ICPL 87, GAUT 82-104, BP 86-34	BP 86-112,GAUt 89-21, GAUT 85-44	BDN -2, GT-1, BP 86-204, GAUT 85-39, GT 100	Field survey
Sorghum	*Oligonychus Indicus*	IS 2312, SR 888-1, CSH-1,	GSH 1, SR 1289, CSH-9, SR 382-1,SR 402	GJ-38, SR-389, GJ 37, GJ 39, GJ 36 and SR 1089	Damage intensity
Chilli	*Polyphagotarsonemus latus*	Jwala, RHRC Errect, AEG-77	Guj.Chilli-2, S.G. 5	G-4, PBS 86-1, Guj. Chilli-1	Population density and resultant leaf curl
Rice	*Steneotarsonemus spinki*	Masuri,GR-10, IR-50,GR-3	GR-11,GR101,IR-28,GR-6	Jaya, Gurjary, IR-21, GR-6	Population density and damage intensity

(continued)

Table 10.6 (continued)

Crop	Mite pests	Susceptibility			Mode of evaluation
		Less	Moderate	High	
Carnation	*Tetranychus urticae*	Orange firato	Gaudina, Flanal, Yellow dotcom	Beaumonde	Population density and damage intensity
Rose	*Tetranychus urticae*	Sakira	Naranga	Gold strike, Noblesse, Passion, First red	Population density and damage intensity
Chrysanthemum	*Tetranychus urticae*	Jaya, IIHR-6	Shyamal, Silk brocade	Ravi kiran, CS-16, Yellow gold, Red gold, Flirt, Nilima	Population density and damage intensity
Sorghum	*Oligonychus indicus*	SR 2859, SR 2820, SR 2879, SR 2868	SR 2706, SR 2821, GJ 35, SR 2865, SR 2872, SR 2869	GJ 42, SR 2877, SR 2824, GJ 38	Population density and damage intensity
Mango	*Tetranychus fijiensis*	Nileshan, Neelphanso, Vashi-badami, Vanraj	–	Kesar, Alphanso, Totapuri	Population density and damage intensity
Gerbera	*Tetranychus urticae*	Cherany	C.F. Gold, Stanza	–	Population density and damage intensity
French bean	*Tetranychus urticae*	PDR-14, HUR-15, IPR-98-31	HUR-137, HUR-203	Suvidha, HUR-203, Arka Aroop, Arka	Population density and damage intensity

Brinjal	*Tetranychus urticae*	GBL-4, NSR-1, AB-08-05, Arkakeshav	GJB-2, Panjabbasant, Pant bahar	GBL-1, GBL-2, GBL-3, GOB-1, NSRP-1, NSRP-2, JBGR-1, Arkanidhi, Pusaupkar	Population density and damage intensity
Sorghum	*Oligonychus indicus*	SR 2859, SR 2820, SR 2879, SR 2868	SR 2462, SR 2819, SR 2873, SR 2871, DJ 6514, SR 2853	GJ 42, SR 2877, SR 2824, GJ 38	Population density and damage intensity
Carnation	*Tetranychus urticae*	Orange, firato	Gaudina, Flanal, Yellow dotcom	Beaumonde	Population density and damage intensity
Chrysanthemum	*Tetranychus urticae*	Jaya, IIHR-6	Shyamal, Silk brocade	Ravi kiran, CS-16, Yellow gold, Red gold	Population density
Rice	*Steneotarsonemus spinki*	Masuri, IR-28	GR-101, GNR-3, GR-104, GR-102, GAR-1, GR-7 and GR-11	Gurjari and Jaya	Population density
Mango	*Tetranychus fijiensis*	Nileshan, Neelphanso, Vashi-badami, Vanraj	–	Kesar, Alphanso, Totapuri	Population density
Carnation	*Tetranychus urticae*	Domingo	Famosa, Cherry Solar	Rubisco	Population density
Sapota	*Tuckerella kumaonensis*	Zumakhiya	–	Cricket Ball, Kalipatti, Murabba, Challa Collection-3, Paria Collection	Population density

Table 10.7 Varietal preference based on development of phytophagous mite on different cultivars/host plants under laboratory condition

Crop	Mite pests	Preference		
		Less	Moderate	High
Okra	*Tetranychus macfarlanei*	AE 36-2, Perkin's Long Green, AE 100	G.Okra-1, Pusa Padmini, Pusa Sawani, Sultan's Improved Sawani	Selection 2
Brinjal	*Tetranychus urticae*	ABH-2, Ringan Gulabi, Doli -5	PLR-1, M-62, DBR-8	Morvi 4-2,ABH-1, M-36
Vegetable crops	*Tetranychus ludeni*	–	Brinjal	Indian bean, little gourd, cowpea, okra
Sorghum	*Oligonychus indicus*	SR 382-1,JS 2312,SR 888-1,SR 389	GJ-36, SR 1088, SR 1298	GJ-38, GJ-37, GJ-35, SR-993, GJ-40, SR-1289, SR-705

Table 10.8 Acaricides used in the field against mite pests

Type of acaricide	Mode of action	Acaricides; R = resistance evidence in T. urticae
Carbamates	Acetylcholinesterase inhibitors	Aldicarb*, bendiocarb, carbaryl, carbofuran, fenobucarb, formetanate*, methiocarb*, oxamyl, propoxur*
Organophosphates		azinphos-ethyl*, azinphos-methyl*, chlorfenvinfos, chlorpyrifos*, chlorpyrifos-methyl, coumaphos*, demeton-S-methyl*, diazinon*, dichlorvos*, dimethoate*, disulfoton*, ethion, fenthion, heptenophos, malathion*, mecarbam, methamidophos, mevinphos, monocrotophos*, omethoate, parathion*, parathion-ethyl*, phorate, phosalone*, phosmet, phoxim, pirimiphos-ethyl, profenophos*, quinalphos, sulfotep*, tetrachlorvinphos, triazophos*, trichlorfon, vamodothion*
Cyclodiene Organochlorines	GABA-gated chloride channel Antagonists	Endosulfan*, dicofol*
Organotin miticides Benzoylureas	Synaptic agonists	Acrinathrin*, bifenthrin*, cyhalothrin*, cypermethrin*, fenpropathrin*, fenvalerate*, flucythrinate*, flumethrin*, tau-fluvalinate*, halfenprox*, lubrocytrhinate, permethrin*
METI-acaricides	Chitin synthesis inhibitors	Flucyloxuron, flufenoxuron
Spirocyclic compounds	Mitochondrial complex I electron transport inhibitors	Fenazaquin*, fenpyroximate*, pyrimidifen*, pyridaben*, tebufenpyrad*
Organochlorine	Inhibitors of acetyl coenzyme A carboxylase	Spirodiclofen*, spiromesifen
Hydrazine carbazate	Unknown or uncertain mode of action	Dicofol*, tetradifon*
Macrocyclic Lactones	Chloride channel activators	Abamectin*, milbemectin*
N-substituted halogenated pyrrole	Uncoupler of oxidative Phosphorylation	Chlorfenapyr*
Other chemicals	Inhibitors of mitochondrial ATP Synthase	Diafenthiuron
	Octopamine antagonist	Amitraz*
	Mitochondrial electron transport Inhibitors	Acequinocyl*, fluacrypyrim*
	Mite growth inhibitors	Clofentezine*, hexythiazox*, etoxazole *

Source: IRAC (2008), Van Leeuwen et al. (2009) *Report of resistance

Table 10.9 List of mite pests on different crops, their field identification, damage symptoms, period of occurrence and pest status in India

S. No.	Name of mite pest (common name)	Crops attacked	Field identification	Pest status	Damage symptoms status	Period of occurrence	Distribution in India
1	*Tetranychus cinnabarinus*	Vegetables Cucurbitaceous crops	Carmine mites (reddish brown) with two black spots on either side of the body	Major	Yellowish spots; those increase to form yellow patch; leaves fall off; plants often are covered with dense webs affecting normal physiological activities; in case of heavy infestation, plants may die	January–April	Central India
2	*Tetranychus urticae*	Vegetables, ornamental plants, oilseeds, pulses, plantation crops, fibre crops, fruit trees	Reddish brown with two black spots on either side, mostly occur on undersurface of leaves	Major		Throughout more during summer months	Cosmopolitan first appear
3	*T. neocaledonicus* (red vegetable mite)	Vegetable, ornamental plants, pulses	Body carmine reddish, nymphs green with dark specks, on undersurface of leaves	Major	Yellowish spots; those increase to form yellow patch; leaves fall off; plants often are covered with dense webs affecting normal physiological activities; in case of heavy infestation, plants may die	Throughout more during summer months	Cosmopolitan first appear
4	*T. ludeni* (carmine mite)	Vegetables	Carmine body with red legs, occur on undersurface of leaves	Major	Yellowish spots; those increase to form yellow patch; leaves fall off; plants often are covered with dense webs affecting normal physiological activities; in case of heavy infestation, plants may die	Throughout more during summer months	Cosmopolitan first appear

5	*T. macfarlanei*	Vegetables fibre crops	Reddish brown light brown with red legs on undersurface of leaves	Minor	Yellowish spots; those increase to form yellow patch; leaves fall off; plants often are covered with dense webs affecting normal physiological activities; in case of heavy infestation, plants may die	September to February	Central and South India
6	*Tetranychus spp.*	Vegetables, Cucurbits, Ornamental	–	Minor	Yellowish spots; those increase to form yellow patch; leaves fall off; plants often are covered with dense webs affecting normal physiological activities; in case of heavy infestation, plants may die	Summer	Central India
7	*Eutetranychus orientalis* (oriental citrus mite)	Fruit trees ornamental plants shade trees	Brownish greenish Brown! Black body, mostly on upper surface of leaves	Major	Leaves become yellow and then turn brown; plants defoliate, often covered with thin webs	September to February	Cosmopolitan
8	*Eotetranychus sp.*	Cabbage, Papaya	–	–	–	–	–
9	*Eotetranychus broodryki*	Pigeon pea	Green mite, Body linear	Minor	–	September–February	Central India
10	*Eotetranychus rajouriensis*	Ber (*Ziziphus* spp.)	Green mite	M	–	–	–
11	*Schizotetranychus andropogoni*	Cereals, fodder commercial crops	Pale body with dusky spots on undersurface (colonial mite)	Minor	Whitish patches appear on either side of mid-rib, in longitudinal rows	Premonsoon and post-monsoon	All over India

(continued)

Table 10.9 (continued)

S. No.	Name of mite pest (common name)	Crops attacked	Field identification	Pest status	Damage symptoms status	Period of occurrence	Distribution in India
12	*S. hindustanicus*	Fruit trees	Flat, light green mile on both surfaces of leaves	Minor	Yellowish stipplings on leaves	Premonsoon	South India, East India
13	*S. cajani*	Pulses	Yellowish mite	Minor	Yellowish stipplings on leaves	Premonsoon	Eastern India
14	*Panonychus citri* (citrus red mite)	Fruit trees	Dark red, dorsal setae very long set on tubercles on	Minor	Infested leaves become speckled later turn greyish	Premonsoon	Eastern & Northern India
15	*P. ulmi* (European red mile)	Fruit trees	Reddish	Major	Leaves turn yellow, brown, scorchy	–	Northern India
16	*Panonychus sp.*	Banyan tree	–	–	–	–	–
17	*Petrobia latens* (brown wheat mite)	Cereals, spices	Dark brown, greenish brown with pale yellow legs, on undersurface of leaves	Major	Leaves dry up from tip downwards, yellow spots on leaves; later those leaves turn bronzy	December to April	Western and Northern India
18	*Petrobia sp.*	Nerium (ornamental plant)		Minor			Central India
19	*Bryobia praetiosa* (brown mite)	Fruit trees	Dark brown or greenish brown with fan-like setae dorsally, occur on upper surface	Minor	Leaves turn brownish plants defoliate	Spring	North India
20	*Oligonychus indicus* (sugarcane red spider)	Commercial crops, fodder plantation crops, fruit trees, cereals	Greyish green with black blotches, occur on undersurface of leaves	Major	Reddish patches appear on sugarcane, white patches on maize, paddy, banana	September–October	All over India

21	*0. coffee* (tea red spider)	Plantation crops	Bright crimson dorsally, bronzy ventrally, occur on upper surface of leaves	Major	Leaves turn copperish brown	April–June	Northeast India, South India
22	*O. punidae*	Fruit trees	–	Minor	Brownish leaves with castoff skins	–	Northern India
23	*O. mangiferus* (mango red spider)	Fruit trees	Dark red dorsolaterally, pale red mediodorsally, occur on upper surface of leaves	Minor	Infested leaves brownish, later fall off	Throughout the year	All over India
24	*0. oryzae* (paddy mite)	Cereals	Whitish mite	Minor	Whitish spots on upper surface of leaves	Premonsoon	South India
25	*0. biharensis*	Fruit trees, shade trees	Greyish mite	Minor	Whitish spots on leaves	Premonsoon	South India
26	*Eutetranychus hirsti* (fig mite)	Fruit trees	Greenish yellow with black blotches, occur on undersurface of leaves	Minor	Transparent green patches appear on leaves, heavy defoliation	May–October	Northern India
27	*Brevipalpus phoenicis* (scarlet mite)	Fruit trees, plantation crops, ornamental plant species	Dark reddish flat mite with black or green spots, occur on undersurface of leave	Major	Infested leaves turn pale yellow and develop brownish patches	All throughout the year	All over India
28	*B. californicus* (citrus flat mite)	Fruit trees	Body with deep reddish pattern; occur on undersurface of leaves	Minor	Infested leaves produce brownish patches	All throughout the year	All over India

(continued)

Table 10.9 (continued)

S. No.	Name of mite pest (common name)	Crops attacked	Field identification	Pest status	Damage symptoms status	Period of occurrence	Distribution in India
29	*B. obovatus*	Fruit trees, plantation crops	Body dark red with dark pigmentation; occur on the undersurface	Minor	Produce chlorotic patches with concentric rings of reddish resinous material	All throughout the year	All over India
30	*Brevipalpus sp.*	Pigeon pea, ber, safed jamun, croton, tagod-wild plant	–	–	–	–	All over India
31	*Raoiella indica* (coconut red mite)	Fruit trees, oilseed	Reddish round mite, occur Minor on undersurface of leaves	Minor	Infested leaves show reddish spots	January March	All over India
32	*Raoiella macfarlanei*	Jamun	–	–	–	–	–
33	*Tenuipalpus sp.*	Citrus plant	–	–	–	–	–
34	*Pentamerismus sp.*	*Nerium* sp.	–	–	–	–	–
35	*Dolichotetranychus floridanus* (pineapple sheath mite)	Fruit trees	Elongated orange mite. On undersurface of leaves	Minor	Feeding at basal part of leave causes rust-like spots	March–June	North East India
36	*Larvacarus transitans* (ber gall mite)	Fruit trees	Elongated body with three pairs of legs	Minor	Produce round galls on ber	May–June	Eastern and Western India
37	*Aceria mangiferae* (mango bud mite)	Fruit trees	Whitish worm-like mite, within buds	Major	Incriminated to causing vegetative and floral malformation	All throughout year	All over India
38	*Aceria cajani* (pigeon pea sterility mosaic mite)	Pulses	Do occur on undersurface	Major	Causing sterility mosaic disease	Mid-September–February	Southern Western and Eastern India

39	*A. lycopersici* (brinjal erineum mile)	vegetables	Do occur on undersurface	Minor	Causes erineum (excessive growth of hairs) on leaves	All throughout year	All over India
40	*A. litchii* (litchi erineum mite)	Fruit trees	Light brownish mite. Occur on undersurface	Major	Producing velvety chocolaty erineum	June–July	Eastern India
41	*A. guerreronis* (coconut eriophyid mite)	Oilseeds	Light brownish, occur underneath perianth	Major	Triangular patches appear close to pre-old nuts; damaged fruits become shriveled, with brownish patches; nut deformed; longitudinal splits appear; nuts fall	Throughout the year	South India
42	*Aceria sacchari* (sugarcane blister mite)	Commercial crops	Creamish body	Minor	Cause red blisters on inner surface of leaf sheath; such leaves gradually wither	–	North and South India
43	*Aceria jasmini* (Jasmine erineum mite)	Ornamental crops	Whitish body	Minor	Cause red blisters on inner surface of leaf sheath, such leaves gradually wither	–	North and South India
44	*Aceria ficus* (fig eriophyid mite)	Fruit trees	Do	Minor	Vector of fig mosaic disease	Throughout the year	North India
45	*Acaphylla theae* (tea pink mite)	Plantation crops	Spindle-shaped robust body, occur on both surfaces	Major	Infested leaves become pale, leathery, veins and margins show pinkish discolouration	All throughout the year	South and Northeast India
46	*Calacarus carinatus* (tea purple mite)	Plantation crops	Purple-coloured mite with 5-waxy dorsal longitudinal ridges	Major	Infested leaves become copperish brown discoloration ultimately turn purplish bronze	All throughout the year	NE and South India

(continued)

Table 10.9 (continued)

S. No.	Name of mite pest (common name)	Crops attacked	Field identification	Pest status	Damage symptoms status	Period of occurrence	Distribution in India
47	*Aceria gossypii*	Cotton	Whitish worm like	–	–	–	–
48	*Cisaberoptus kenyae* (mango web mite)	Fruit trees	Whitish body	Minor	White silvery coaling on upper surface of leaves under which mites live	All throughout the year	Western and Northern India
49	*Calepitrimerus azadirachta*	Neem	Whitish worm like	–	–	–	–
50	*Eriophyes cernuus* (ber woody gall mite)	Fruit trees	Light brownish mite	Minor	Produced woody galls on ber	All throughout the year	Western and Northern India
51	*Phyllocoptruta oleivora* (citrus rust mite)	Fruit trees	Do	Minor	Produce silvery skin on fruits which later turn blackish/chocolaty brown on ripe fruits. Such rusty fruits drop prematurely	All throughout the year	Western and Northern India
52	*Polyphagotarsonemus latus* (yellow mile)	Fruit trees, vegetables, ornamental plants, fibre crops, pulses, plantation crops, cereals, spices	Yellowish glossy mite fast moving mostly on young leaves	Major	Infested leaves become crinkled and curled from margin; later, such leaves turn bronzy	All throughout the year	All over India
53	*Steneotarsonemus spinki* (rice sheath mite)	Cereals	Whitish, occur on upper surface of leaf sheath and basal part of leaf blade	Minor	Curling of leaf, emergence of ear heads from panicles malformed, grains deformed and chaffy, ill filled, discoloured grains	Occurrence maximum during booting stage and milk grain stage	Eastern India

54	*S. bancrofti*	Commercial crops	Light brownish	Minor	Internodes give scabby corroded appearance, transparent depressions appear on young stalks	–	Northern India
55	*Tuckrella sp.*	Sapota fruit	Reddish, flat mite having fan-like setae	Minor	–	Sept to March	Central India

Table 10.10 List of predatory mites, host plants and prey mites

No.	Predatory mites	Host plant	Mite host
Family: Phytoseiidae			
1.	*Amblyseius cucurbitae* (Rather)	All cucurbit crops	*Tetranychus urticae, eriophyid mite (?)*
2	*Amblyseius herbicolus* (Chant)	Hollyhock, hibiscus (Jasod)	*T. urticae*
3.	*Amblyseius largoensis* (Muma)	Tomato, marigold, castor, fig	*T. urticae, Eutetranychus orientalis, Aceria lycopersici*
4.	*Amblyseius orientalis* (Ehara)	Citrus group, castor	*E. orientalis*
5.	*Amblyseius raoiellus* (Denmark & Muma)	Oil palm, banana	*Oligonychus indicus, Raoiella indica*
6.	*Amblyseius nucifera* (Gupta)	Coconut, areca palm	*Eriophyid mite (?)*
7.	*Amblyseius (Euseius) alstoniae* (Gupta)	Brinjal, tomato, French bean, beans, okra, mulberry, coriander	*T. urticae, T. cinnabarinus, T. neocaledonicus, O.biharensis*
8.	*Amblyseius (Euseius) bambusae* (Ghai & Menon)	Bamboo	*Spider mite (?)*
9.	*Amblyseius (Euseius) coccineae* (Gupta)	Mango, ramfal, sitafal	*O. mangiferae, T. fijiensis, eriophyid mite (?)*
10.	*Amblyseius (Euseius) coccosocius* (Ghai & Menon)	Marigold, sugarcane	*T. urticae, O. indicus, Aceria saccheri*
11.	*Amblyseius (Euseius)eucalypti* (Ghai & Menon)	Eucalyptus saplings	*Unknown tarsonemid mite*
12.	*Amblyseius (Euseius) finlandicus* (Oudemans)	Okra, chilli, French bean, citrus group, tomato, sorghum, cowpea	*Tetranychus urticae, P. latus, E. orientalis, O. biherensis, A. lycopersicse*
13.	*Amblyseius (Euseius) ovalis* (Evans)	Chilli, til	*Polyphagotarsonemus latus*
14.	*Amblyseius (Euseius) sacchari* (Ghai & Menon)	Sugarcane, baru, sorghum	*O. indicus, Schizotetranychus andropogoni, A. saccheri*
15.	*Amblyseius (Neoseiulus) aceriae* (Gupta)	Coconut	*Aceria guerreronis, R. indica*
16.	*Amblyseius (Neoseiulus) baraki* (Athias-Henriot)	Coconut	*R. indica, O. indicus*
17.	*Amblyseius (Neoseiulus)*	Haldi	*Unknown spider mite (?)*

(continued)

Table 10.10 (continued)

No.	Predatory mites	Host plant	Mite host
	cucumeris (Oudemans)		
18.	*Amblyseius (Neoseiulus) cynodonae* Gupta	Grasses	*O. indicus, O. oryzae*
19.	*Amblyseius (Neoseiulus) fallacis* (Garman)	Chilli, rose	*Polyphagotarsonemus latus*
20.	*Amblyseius (Neoseiulus) indicus* (Narayanan & Kaur)	Sorghum, baru, grasses	*O. indicus*
21.	*Amblyseius (Neoseiulus) longispinosus* (Evans)	Okra, chilli, French bean, citrus group, tomato, sorghum, cowpea, grasses, ramfal, beans, pigeon pea	*T. urticae, T. neocaledonicus, E. orientalis, O. indicus, A. lycopersici, E. broodryki*
22.	*Amblyseius (Neoseiulus) paspalvorus* (De Leon)	Cowpea	*T. ludeni, Oligonychus sp.*
23.	*Amblyseius (Paraphytoseius) multidentatus* (Swirski & Shechter)	Greengram, Blackgram	*T. ludeni,*
24.	*Amblyseius (Typhlodromalus) eucalypticus* (Gupta)	Eucalyptus nurseries	*Unknown tarsonemid mite*
25.	*Amblyseius (Typhlodromalus) ficusi* Gupta	Ornamental ficus	*Unknown spider mite, Aceria sp.*
26.	*Amblyseius (Typhlodromalus) mangiferae* (Chatterjee & Gupta)	Mango	*Oligonychus mangiferae, Tetranychus fijiensis, A. mangiferae*
27.	*Amblyseius (Typhlodromalus) sorghumae* (Gupta)	Sorghum, baru, pearl millet	*O. indicus*
28.	*Amblyseius (Typhlodromalus) arecae* (Gupta)	Arecanut (ornamental)	*Raoelia indica*
29.	*Amblyseius (Typhlodromi) guajavae* (Gupta)	Guava, citrus, papaya	*Brevipalpus sp., Tenuipalpus sp., T. urticae, E. orientalis*
30.	*Amblyseius (Typhlodromips) tetranychivorus* (Gupta)	Gerbera, orchids, spider lily, carnation, gaillardia	*T. urticae, Tenuipalpus sp.*
31.	*Indoseiulus eharai* (Gupta)	Rose, carnation, tomato, jasmine	*T. urticae, Aceria jasmine*

(continued)

Table 10.10 (continued)

No.	Predatory mites	Host plant	Mite host
32.	*Indoseiulus ricini* (Ghai & Menon)	Castor	*Eutetranychus orientalis*
33.	*Phytoseius (Phytoseius) jujube* (Gupta)	Ber	*Green mite*
34.	*Phytoseius (Phytoseius) swirskii* (Gupta)	Chrysanthemum, rose, gerbera	*T. urticae*
35.	*Typhlodromus (Amblydromella) chrysanthemi* (Gupta)	Soybean, maize, sorghum	*Oligonychus indicus, T. urticae*
36.	*Typhlodromus (Amblydromella) mori* Gupta	Mulberry, teak saplings	*T. urticae, eriophyid mite (?)*
37.	*Typhlodromus (Amblydromella) divergentis* (Chaudhri, Akbar & Rasool)	Bamboo, banana, carrot weed, motha, kaner	*Unknown spider mite, Tenupalpid mite, R. indica*
Family: Stigmaeidae			
38	*Agistemus sp.*	*Sesamum*	
39.	*Agistemus fleschneri*	*Lantana* sp.	*Tetranychus sp.*
	Family: Tydeidae		
40	*Pronematus fleschneri*	Okra, tomato, brinjal	*T. macfarlanei, P. latus,*
41	*Tydeus sp.*	Mango	*A. mangiferae*
Family: Erythreidae			
42	*Leptus sp.*	Castor	
Family: Amerosiidae			
43	*Neocypholaelaps stridulans*	Coconut	*Aceria spp.*

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Rodents

11

Mani Chellappan

Contents

Abstract

Rodents are the largest order under the class Mammalia with more than 2000 living species in 30 extant families and as high as 481 genera. Two-thirds of all living rodents belong to one single family, Muridae. Rodents are immensely diverse, ecologically, and they occupy any vacant places on Earth and eat anything. They are an important link in the food web and a number one vertebrate pest and also transform landscapes. Their adaptation is awesome; they adapt to any habitat, any food and any situation. Commensal rodents not only occupy the human habitation, causing economic losses to various stored products of human beings. They also act as vectors of numerous dreaded rodent-borne diseases in human beings. Rodent population dynamics depends on the availability of resources and environmental conditions. The assessment of rodent population

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M. Chellappan (✉)
AINPVPM, Department of Agricultural Entomology, College of Horticulture, KAU, Thrissur, Kerala, India

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in the wild is tricky as most of the pest rodents are nocturnal or subterranean. Pest rodents cause damage to agricultural crops, forestry, poultry industry, aviation and public health sectors. Being a mammal, rodents are extremely intelligent, and because of some limitation in their physiology, rodents are always suspicious. Neophobia and bait shyness are some of the traits which limit the use of rodenticides successfully. As one of the prolific breeders, rodents perform reproductive bounce after a successful control programme. Numerous management methods, viz. environmental, physical and chemical techniques, are being followed to contain the rodent population to a certain extent in a particular habitat.

Keywords

Rodent · Rat · Mouse · Diversity · Assessment · Management · Rodenticide

11.1 Introduction

Rodents (from the Latin *rodere*, ‘to gnaw’) are the quadruped mammals. They are one of the persistent vertebrate pests causing enormous damages to agricultural crops and storage facilities either by direct feeding or by indirect damages, viz. contamination, spillage, spoilage and hording.

Rodents are a dominant group of mammals and the order Rodentia, the largest order of mammals, comprising of 2277 species, 481 genera in 30 extant families (Wilson and Reeder 2005; Macdonald et al. 2015). Family Muridae has two-thirds of all living rodent species under Rodentia. Ecologically, rodents are incredibly diverse. Some species are entirely terrestrial or arboreal, whereas others never emerge out from underground burrows; some species are essentially aquatic, while others are specialists in living in extreme conditions of deserts. Majority of the rodents are omnivorous, while some have specialised food habits like feeding on only a few species of invertebrates or fungi. Size of the rodents also varies; some are very small (pygmy mice, weighing only 5 g), while others are robust (capybaras, the largest South American rodent having a body weight over 70 kg). However, all rodents share one common character, the highly specialised dentition for gnawing, despite their enormous morphological and ecological diversities. In general, rodents have a pair of ever-growing upper and lower incisors, followed by diastema (a gap in the dentition), and molars or premolars; no rodent has canine teeth. The characteristic gnawing helps the rodents to do self-sharpening of their incisors by grind against each other which wears off the softer dentine and makes the enamel edge as chisel blade-like. The gnawing habits and the powerful incisors help the rodents to survive in any environment and hold the key for their success.

Rodents occupy a wide range of natural habitats, including forests, grasslands, the agricultural landscapes and human habitations. They are the most important link in the food chain and food web as a primary consumer and prey for higher animals. The rodent stomach can vary from a simple sac in the dormice (Gliridae – the only rodents without a caecum) to the complex ruminant-like organ of the lemmings (*Lemmus* spp.).

Life history strategies can be short and prolific, as in the *r*-selected house mouse, *Mus domesticus*, or long with low fecundity as in the *K*-selected African spring hare, *Pedetes capensis*, which produces only one progeny each year. Rodent social systems include monogamous water voles (*Arvicola amphibius)*, polygynous wood mice (*Apodemus sylvaticus)* family groups of alpine marmots (*Marmota marmota)* herds of capybara (*Hydrochoerus* sp); and, a very unique among mammals, a blend of monogamy and communal denning in mara (*Dolichotis patagonum)*.

Rodent breeding systems include the single-sex litters of wood lemmings, *Myopus schisticolor*, and the manipulated sex ratio of coypus, where selective abortion of male-biased litters is done by female parents in poor environmental conditions (Gosling 1986). Naked mole rats, *Heterocephalus glaber*, are unique among mammals in the degree of their eusociality (Jarvis 1981) with only one breeding female within the group.

Added to the diversity within the order, there are special adaptations in many individual species and behavioural flexibility of individuals. Thus, brown rat, *Rattus norvegicus*, and house mouse (*Mus musculus*) can be found throughout the world, using their generalist body plan to feed and breed wherever humans go and their sophisticated behaviour patterns to avoid the most cunning and increasingly sophisticated attempts to eradicate them.

Brown rats and house mice, along with the roof rat, *Rattus rattus*, are known as commensal ('sharing the table'; *mensa*: a table, in Latin) rodents, meaning that they are usually found in association with human habitation. However, as the word commensal implies no damage to the host, these rodents might more precisely be termed as kleptoparasitic. Because of the importance of the first two species in medical and experimental psychological research, knowledge of rodent biology is heavily biased on these commensal rodents.

Rodents generally have very acute senses of smell, hearing, touch and taste (Broadford 2015). Social odours play an enormous role in rodent biology, both through a direct impact on behaviour and through the physiological impact through primer pheromones (Johnston 2003). Functional odours are produced in the urine and faeces and in secretions from apocrine and sebaceous glands (e.g. flanks, prepuce, eyes). Scent marking plays an important role in territoriality in many species, and territoriality can affect rodent control.

Olfaction is also important in transferring information between individuals and can affect rodent control. Taste, mediating food preferences and recognition, affects the efficacies of poison baits. The inability of rodents to taste certain compounds at a concentration that is repugnant to humans (e.g. Bitrex® – denatonium benzoate) is used to 'safen' modern rodenticide baits.

Many rodents produce ultrasounds (i.e. sounds above the normal level of human hearing, 20 kHz), which are apparently relevant in courtship and aggression, in eliciting parental care, as alarm signals, and, possibly in echolocation. Sounds in the 'audible' frequencies are also used for these purposes. Hearing is often the first sense to detect the approach of a potential predator; the most extreme case is the middle ear of desert-living kangaroo rats (Dipodidae), which amplifies the movement of the eardrum 92 times, compared with 18 times in humans, meaning that their hearing is four times more superior than ours (Webster 1965).

Touch is a highly developed sense in many rodents: rats and mice with trimmed or removed vibrissae (whiskers) become subordinate when grouped with intact conspecifics. Tactile hairs are found all over the pelage (fur) and are important in ensuring that the rodent moves in close proximity to vertical surfaces, a behaviour that may limit the possible avenues of attack of predators. Closely related to the sense of touch is that of 'muscle awareness' or kinaesthesis, by which a rodent is aware of its physical environment through a combined memory of movement and touch. This is vital for quick escape from predation, where a rodent will run along a 'prerecorded' path at a great speed.

Another important adaptation of rodents is their ability to swim. The brown rat can swim for 72 h nonstop and has been known to enter houses through lavatory U-bends. Commensal rodents can climb brick walls with comparative ease. Other species are accomplished jumpers, with the African spring hare covering 2 m in a single bound. Flying squirrels have membrane on both sides of the body, and one species has been observed to use flapping movements to reach a point that was 1 m higher than its launch pad and to glide a horizontal distance of 135 m (Hanney 1975). A rat or mouse can generally enter any hole through which its head will fit, with young mice being able to enter a gap less than 10 mm high (Meehan 1984).

Rodents also play an important role as environmental engineers, aerating the soil by their extensive burrowing activities. They also change, for instance, the entire nature of the landscape (e.g. beavers). Rodents also help in the dispersal of seeds and, in some cases, aid in spread of pollen.

11.2 Rodent Anatomy

Like any other mammals, rodents share most of the characteristics of class, Mammalia. Rodents are homoeothermic, and body temperature is regulated by heat generated through metabolic processes. With the subcutaneous fat and fur all over the body and by regulating blood flow to the skin, panting and sweating, rodents regulate the body temperature. Rodents are viviparous like other mammals, and the young ones are fed with the mammary glands.

11.2.1 External Morphology

Rodent body has four positions or regions, viz. anterior (cranial), posterior (caudal), dorsal and ventral (Fig. 11.1).
Anterior (cranial) position: Frontal position or the front side of the rodent.
Posterior (caudal) position: Position towards tail or posterior end.
Dorsal side or dorsal position: Upper side of the body.
Ventral sides of rodent body: Under body surface or belly of the rodent.

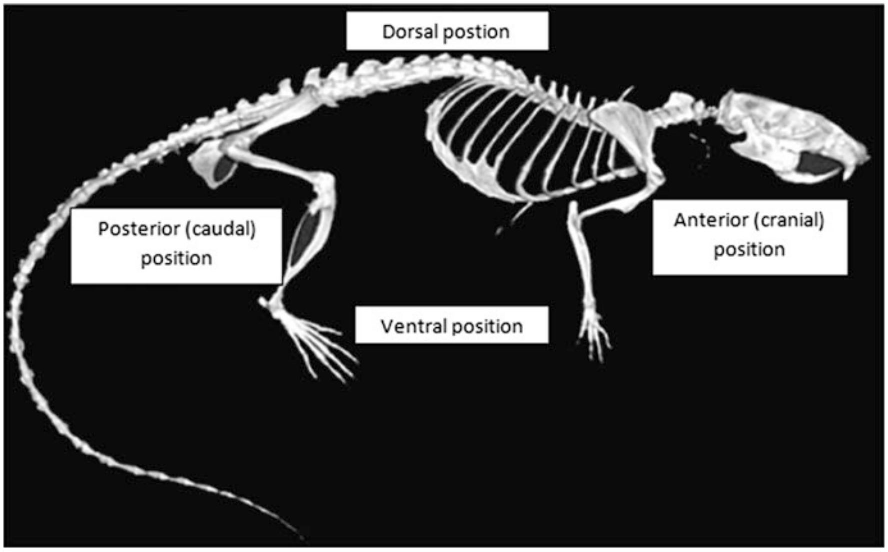


Fig. 11.1 Position of rodent body

Medial and lateral sides:

It is the middorsal longitudinal line passes from the anterior end to the posterior end of the body and divides the body into two-halves, the right and left.

Distal and proximal positions:

Depending on the position of the structures, it may be closer (proximal) and farther (distal) to the reference point.

Rodent body is generally covered with fur and is divisible into head, neck, trunk and tail (Figs. 11.2 and 11.3).

Head: Head is broader posteriorly and tapers anteriorly as a naked muzzle or snout. A pair of nostrils, shaped like inverted commas, is present above the mouth opening, which leads into nasal passages. Below the nostrils is the cleft upper lip, which exposes the two upper incisors. On the lateral sides of the head are large, paired popping eyes. Eyelids have very fine and short eyelashes; the nictitating membrane is reduced. The head bears a pair of external ear or pinna at its postero-lateral position. The mouth is subterminal and located beneath the nostrils and remains guarded by upper and lower lips. Long, stiff, bristle-like hairs, known as *pili lactiles* or vibrissae, are present on both sides of nostrils. They help the animal in measuring width of area through which the animal is to pass even in pitch dark.

Neck: It is a short connective between head and trunk. With the help of neck, the animal can bend its head in different directions.

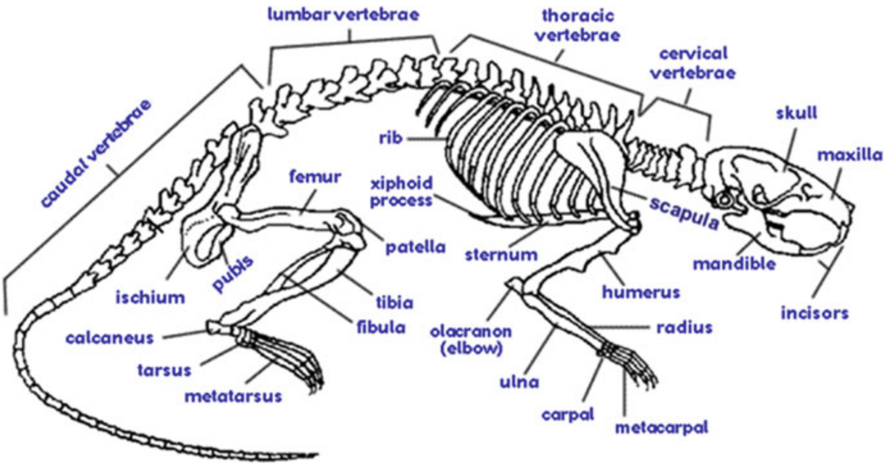


Fig. 11.2 Rodent skeleton

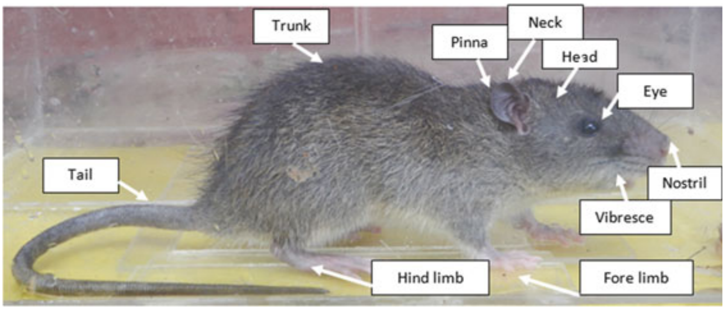


Fig. 11.3 External morphology of a rat: lateral view

Trunk: It is the major part of the body, which has two parts – anterior narrow but stouter thorax and posterior wider softer abdomen. The ventral surface of female bears six pairs of teats or nipples (three pectoral/thoracic and three inguinal/abdominal). The trunk bears two pairs of limbs, two forelimbs and two hindlimbs. Forelimbs are smaller than the hindlimbs. Five digits are present in each limb. The first digit is thumb or pollex, which is much reduced with a peculiarly flatted nail and two phalanges. Nail is keratinised structure occupying position above the distal phalanx of each digit. Typical walking pads, the tori, are present on the tips of digits, palm and the base of palm. These are also present on the feet, but palms and soles do not have hairs. Anus lies posterior-ventrally at the base of the tail.

Tail: It is long and cylindrical and tapering structure present above the anus. It bears overlapping scales and sparse hairs in between. Tail is used as a balancing organ for the animal while trotting or climbing or swimming.

11.2.2 Internal Anatomy

Rodent body is completely invested in fur-covered skin, and it consists of two layers, viz. the epidermis (outer skin) and the dermis (inner skin).

(a) **Outer skin:** It is made up of multiple layers of cells, called stratified squamous epithelium. It consists of five principal layers, viz. germinative layer or stratum germinativum, prickle cell layer or stratum spinosum, granular layer or stratum granulosum, clear layer of dead cells or stratum lucidum and the outermost layer of skin containing keratinised cell or stratum corneum. There are some layers of cells above the stratum lucidum, which constitute the stratum corneum. The cells of this layer contain keratin and have lost all other cell organelles, including nuclei.

(b) **Dermis:** It develops from the mesoderm of embryo. It is composed of dense fibrous connective tissue with blood vessels, lymph vessels, nerve fibres, pigment cells, etc.

Hairs, cutaneous glands and claws are formed from the skin. Major skin glands are sudoriferous glands (sweat glands), sebaceous glands (oil glands), mammary glands (modified sweat glands), meibomian glands (modified oil glands, present along the edges of the eyelids) and ceruminous glands (wax glands, present in the external auditory canal of external ear).

11.2.2.1 Digestive System

Alimentary canal, associated structures and glands constitute the digestive system.

Alimentary canal: It is a coiled tube having variable diameter at different positions. It begins at mouth and ends at anus. Various parts of the digestive system include the mouth, buccopharyngeal cavity, oesophagus, stomach, small intestine, large intestine and anus:

(a) **Mouth**: The mouth opens in the buccal cavity that is surrounded by the vestibule, which is a space between the lips, cheeks and teeth.

(b) **Buccopharyngeal cavity**: It is space enclosed by two jaws, and it consists of broader buccal cavity in the anterior region and narrow pharynx in the posterior region. Jaws bear teeth. The teeth are heterodont (different sets of teeth), thecodont (base of the tooth is completely enclosed in a deep socket of bone) and monophyodont (only one set of teeth develops in their lifetime). Each jaw carries two ever-growing incisors (growing teeth) and six molars. A sharp cutting edge is maintained due to the absence of enamel on the surface. A space called diastema occurs between incisors and molars due to the absence of canines and premolars. The dental formula of a typical rodent is (1/1, 0/0, 0/0 and 3/3) $\times 2 = 16$. The middle of buccal cavity contains a muscular tongue. Taste buds occur on tongue as well as lining of buccopharyngeal cavity. Pharynx lies behind, and it is a common chamber for the passage of food and air.

(c) **Oesophagus**: It is a short tube situated dorsal to the trachea, and it leads into the stomach.

(d) **Stomach**: It lies on the left side behind the diaphragm. The curvature of the stomach is more on left side compared to right side. Oesophagus opens into the stomach through cardiac orifice/valve, and pyloric sphincter is present at the posterior, where it meets duodenum. Stomach contains goblet cells for mucus, oxyntic (parietal) cells for HCl and peptic cells for pepsinogen secretions.

(e) **Small intestine**: Stomach leads into small intestine, which can be differentiated into three parts: U-shaped duodenum, a straight jejunum and highly coiled ileum. Glands in the small intestine secrete intestinal juices or *succus entericus,* which contains lipase, nuclease, peptidase, lactase, sucrase and maltase enzymes to digest the food.

(f) **Large intestine**: It has three parts – caecum, colon and rectum. Caecum is slightly constricted about its middle. The constriction subdivides the caecum into two parts, the apical and basal portions. The apical portion contains a vermiform appendix. Caecum opens into the colon, the first part of large intestine which leads into rectum and finally opens outside through the anus.

Digestive Glands of Rodent Digestive System

(a) **Salivary glands**: Three pairs of salivary glands are present in rodents. They are:
    (i) Sublingual glands
    (ii) Submandibular glands
    (iii) Parotid glands

(b) **Liver**: Liver is located below the diaphragm in the upper and right side of the abdominal cavity. The liver of rat has four lobes (left, middle, right and caudate), and the spigelian lobe is a part of caudate lobe. The liver cells (hepatocytes) secrete bile which is carried to the duodenum by bile duct. Bile contains no digestive enzymes but helps in digestion of food in the small intestine. Gall bladder is absent in rats, like whales and horses.

(c) **Pancreas**: It is much diffused structure and is present between the duodenal loops. It secretes pancreatic juice, which contains digestive enzymes, such as trypsinogen (proenzyme), amylopsin and lipase. Islets of Langerhans of the pancreases secrete certain hormones, such as insulin and glucagon. Insulin converts glucose into glycogen in the liver and muscles.

(d) **Gastric glands**: These are found in stomach and secrete gastric juice containing digestive enzymes (e.g. pepsin) and hydrochloric acid which help in digestion of food.

(e) **Intestinal glands**: These are present in the small intestine and secrete intestinal juice containing digestive enzymes (e.g. maltase, sucrase, lipase, etc.) which help in digestion of food.

11.2.2.2 Respiratory System of Rodents

It consists of respiratory tract, two lungs and a mechanism for inspiration and expiration. Respiratory tract consists of nostrils, nasal chambers, internal nares (nostrils), glottis, larynx, trachea, bronchi, bronchioles and alveoli. The nostrils lead into the olfactory or nasal chambers. The two nasal chambers lead into pharynx through internal nares. Pharynx contains a slit-like glottis, which leads into voice box called larynx. Larynx passes into trachea and wind pipe which runs ventral to oesophagus. Trachea divides into two primary bronchi that pass into lungs.

The lungs are placed on either side of the heart in the thoracic cavity with a covering of visceral pleura. There are three lobes of the right lung and only one in the left. Each lung possesses a large number of alveoli, where gaseous exchange occurs between air and blood.

11.2.2.3 Circulatory System

Blood vascular and lymphatic systems constitute the circulatory system of rodents:

(a) **Blood vascular system**: Like other mammals, rodents possess closed and double circulation. Blood, heart and blood vessels constitute the vascular system.

Blood: The volume of blood is about 5–7 ml/100 gm body weight. The blood consists of blood plasma and three types of blood corpuscles, namely, red blood corpuscles (6–7 lakh ml^{-3}), white blood corpuscles (6000–10,000/ml^{-3}) and platelets. Mature RBCs are without nucleus. They contain haemoglobin (respiratory pigment). WBCs provide immunity and defence against diseases. The platelets help in clotting of blood.

Heart: The heart lies on the midline and is placed obliquely in the thoracic cavity, surrounded by pericardial cavity.

The heart has four chambers, viz. atrium (right and left) and ventricle (left and right). Blood flows from the right atrium to the right ventricle via the tricuspid valve (right atrioventricular valve) with three cusps of fibrous tissue. Blood flows from the left atrium to the left ventricle via the bicuspid or mitral valve (left atrioventricular valve). Well-developed arterial and venous system similar to other mammals is found in the rat. Only the left aortic arch is present and two precavae are present in the rat. Hepatic portal system is comprised of veins collecting blood from alimentary canal and supplying to the liver after branching in capillaries. The renal portal system is absent.

(b) **Lymphatic system**: Lymph vessels, lymphatic nodes and lymph constitute the lymphatic system. Lymph is a colourless fluid, which is similar to blood but lacks red blood corpuscles and blood platelets. Lymph is formed by lymph capillaries from tissue fluid. Lymph capillaries join to form lymph vessels. At places, lymph vessels bear lymph nodes. The latter contain minute channels where germs are entrapped by leucocytes. Lymph nodes also produce lymphocytes. Tonsils, a type of lymphatic node, are absent. Lymph vessels form lymph ducts of two types, right and thoracic. They also open into veins.

11.2.2.4 Excretory System

The excretory system of rodents includes paired kidney, ureters, a urinary bladder and urethra.

(a) **Kidneys**: There is a pair of dark red and bean-shaped kidneys. The right kidney is slightly higher in position. The kidney consists of outer cortex and inner medulla. A kidney has numerous microscopic functional units called nephrons. Each nephron is made up of a cup-shaped Bowman's capsule, proximal convoluted tubule (PCT), Henle's loop and distal convoluted tubule (DCT). The Bowman's capsule contains a meshwork of blood capillaries, the glomerulus. Filtration of metabolic wastes takes place in the glomerulus. Filtrate comes to the Bowman's capsule from the glomerulus and then to the other parts of the nephron.

(b) **Ureters**: There is a pair of ureters. Each ureter arises from each kidney. Ureters carry urine from the kidneys to the urinary bladder.

(c) **Urinary bladder**: It is muscular sac-like structure in which two ureters open. The urinary bladder is weak and stores urine temporarily, and hence the rats urinate very frequently. The urine thus discharged is useful to mark the territories in case of dominant animal.

(d) **Urethra**: In male, it carries both urine and semen. In female, it carries urine only. Thus, in male rat, there is only one urinogenital aperture to pass urine and semen. However, in female rat, both urinary and genital apertures are separate.

11.2.2.5 Nervous System

The nervous system is divisible into three main parts:

(a) **Central nervous system (CNS)**: The brain and spinal cord comprises the CNS; the brain is within the skull, while the spinal cord is within the vertebral column.

Brain: The brain is covered by three membranes or meninges. The innermost membrane is called pia mater, the next is the arachnoid mater (= arachnoid membrane) and outermost is the dura mater. The subdural space is present below the dura mater, and the subarachnoid space lies below the arachnoid mater. These spaces are filled with a fluid. The meninges are protective in function.

The brain is composed of two large halves or hemispheres. The cerebral hemispheres form the largest part of the brains. The medulla oblongata tapers posteriorly and is inserted into the spinal cord. The spinal cord is a long, tube-like, thick-walled structure that emerges out through the foramen magnum of the skull and passes through neural canal of vertebral column.

(b) **Peripheral nervous system (PNS)**: The cranial (12 pairs) and spinal (33 pairs) nerves arising from the brain and spinal cord, respectively, in rat constitute the PNS.

(c) **Autonomic nervous system (ANS)**: Involuntary actions are being coordinated by the autonomic nervous system. It consists of both sympathetic and parasympathetic nervous systems.

11.2.2.6 Sense Organs

Skin has tangoreceptors (receptors of pressure and touch), thermoreceptors (receptors of temperature), algesireceptors (receptors for pain) and rheoreceptors (receptors for current or vibrations). Gustatoreceptors (taste receptors) occur in the form of taste buds over tongue and posterior part of palate. Olfactoreceptors are located in olfactory epithelium present in nasal chambers. They perceive the sensation of smell. Smell is also perceived by a pair of Jacobson's organs present in the wall of buccal cavity of rat. Organs of sight are eye, while statoacoustic organs are ears.

11.2.2.7 Reproductive System

11.2.2.7.1 Male Reproductive System

A pair of testes, epididymis, vasa deferens, urethra, penis and spermatic cord (Raj 1984, 2015) constitutes the male reproduction organs in rat.

(a) **Testes**: A pair of testes is found in the scrotal sacs. Each testis is an elongated and ovoid body attached posteriorly to scrotal sac by gubernaculum. Testis of male rat descends in the scrotal sacs through inguinal canal when the animal is in between 30 and 40 days of its life. The inguinal canal remains open throughout life, but during sexually inactive period, the testes may be withdrawn into abdominal cavity.

(b) **Epididymis**: These are paired structures. Each epididymis is a mass of long narrow coiled tubule lying along the testis, which consists of anterior caput epididymis, middle corpus epididymis and posterior cauda epididymis. Epididymis stores the sperms.

(c) **Vasa deferentia**: There is a pair of vasa deferentia. A vas deferens arises from the cauda epididymis. Vasa deferentia carry sperms.

(d) **Seminal vesicles**: A paired structure, which is large and lobulated, except for the smooth tip where it is doubled back upon itself. They are not storehouses for sperms. Their secretion is alkaline and forms the bulk of seminal fluid (semen).

(e) **Urethra**: It is divided into three parts:

    (i) Prostatic urethra is surrounded by the prostate gland.
    (ii) Membranous urethra is the shortest portion and runs from the prostate to the bulb (base) of the penis.
    (iii) Penile urethra passes through the penis and opens at the tip of the penis as urinogenital aperture.

(f) **Penis**: It is a copulatory organ covered by a loose sheath, the prepuce. The penis of the rat has a bony process called the os penis (baculum).

Male Accessory Glands:

(a) Ampullary glands: The outer end of the vas deference near the entrance into the urethra is enlarged into ampulla, which contains ampullary glands to secrete mucus.
(b) Vesicular glands: These are branched glands, which originate from the vas deferens behind the ampulla.
(c) Coagulating glands: Closely applied along the minor curvature of the seminal vesicles and within the same sheath are the coagulating glands. The secretion of these glands serves to coagulate the seminal fluid (semen).
(d) Prostate glands: There are two prostate glands, whose secretion is rich in citric acid, lipid and acid phosphatase.
(e) Cowper's glands (bulbourethral glands): These are one pair, which originate from the urethra at the base penis. They produce a secretion during sexual excitement, which protects the sperms from traces of acids found in the urethra (as the urine also passes through the penile urethra).
(f) Prenuptial glands (glands of Tyson): They develop from the skin forming prepuce. They are modified sebaceous (oil) glands, which secrete peculiar odorous secretion.

11.2.2.7.2 Female Reproductive System

It consists of a pair of ovaries, fallopian tubes, uteri, a common vagina and a clitoris:

(a) **Ovaries**: Ovaries are paired small yellowish compact structures suspended in the body cavity by mesovarium.
(b) **Fallopian tubes** (oviducts or uterine tubes): There is one pair of convoluted fallopian tubes. Each fallopian tube begins with fimbriated funnel, which receives ova from the ovary. As the fertilisation is internal, it takes place in the dilated uppermost portions of the fallopian tubes.
(c) **Uterus** (womb): The uterus is a hollow muscular structure. The uterine horns are fused near the vagina. The wall of the uterus consists of outer covering of peritoneum (perimetrium), middle layer (myometrium) and the inner layer of simple columnar epithelium (endometrium). The embryo gets attached to the uterine wall through placenta. Embryonic development takes place in the uterus. Placenta provides the physiological connection between developing foetus and uterine wall (endometrium) of the mother.
(d) **Vagina**: It is a tubular structure, which extends from the uterus and opens outside as vaginal opening (= vulva). Penis of the male rat is inserted into the vagina during copulation. The vagina also helps to deliver the young ones at the time of birth.
(e) Clitoris: It corresponds to the penis of the male, but it is reduced in size and does not have any passage (it is solid structure). The clitoris is found anterior to the vulva.

Female Accessory Glands:

(a) **Vestibular glands**: These are small mucous glands, which open on the surface of the vestibule of the vagina.
(b) **Bulbourethral glands**: These are small glands, which are present in relation with the urethra.
(c) **Preputial glands**: There is one pair of large preputial glands near the tip of the clitoris.

11.3 Rodent Classification

Modern-day rodents are descendants of Paramyidae, insectivore-like ancestor, which arose about 60 million years ago in the late Palaeocene (Wood 1962). The genus *Aplodontia*, the American mountain beaver, is the most ancient surviving lineage of rodents. Great diversity exists in the contemporary rodents as they are very small to gigantic sizes.

11.3.1 Systematic Position

Kingdom: Animalia
Subkingdom: Bilateria
Infrakingdom: Deuterostomia
Phylum: Chordata
Subphylum: Vertebrata
Infraphylum: Gnathostomata
Superclass: Tetrapoda
Class: Mammalia
Subclass: Theria
Infraclass: Eutheria
Order: Rodentia

Order Rodentia is divided into five major suborders (Plate 11.1–11.5) (Editors of Encyclopaedia Britanica 2020; Myers et al. 2020)**:**

1. Anomaluromorpha
2. Castorimorpha
3. Hystricomorpha
4. Myomorpha
5. Sciuromorpha

11.3.2 Suborder Anomaluromorpha

It consists of two families, viz. Anomaluridae and Pedetidae

11.3.2.1 Family Anomaluridae

Family Anomaluridae includes genera *Anomalurus*, *Idiurus*, and *Zenkerella*. Some of the members of this family have the gliding ability (e.g. pygmy and large anomalures) using the fur-covered membrane. Anomalures have slender bodies with large eyes, long limbs and dense silky fur. Large and pygmy anomalures are nocturnal, and they eat bark, oil palm pulp, insect, etc. A flightless anomalure, a rare species, feeds on termites (Plate 11.1).

11.3.2.2 Family Pedetidae

Commonly called as spring hare (*Pedetes capensis*), which lives in open arid habitats and cultivated area of southern and eastern Africa. Spring hare is about a size of a rabbit. It uses its long and powerful hind legs for jumping (2 to 3 metres, i.e. 6.6 to 9.8 feet, when the animal is alarmed). Apart from grass, the spring hares eat agricultural crops and even locust also (Plate 11.1).

11.3.3 Suborder Castorimorpha

11.3.3.1 Family Castoridae

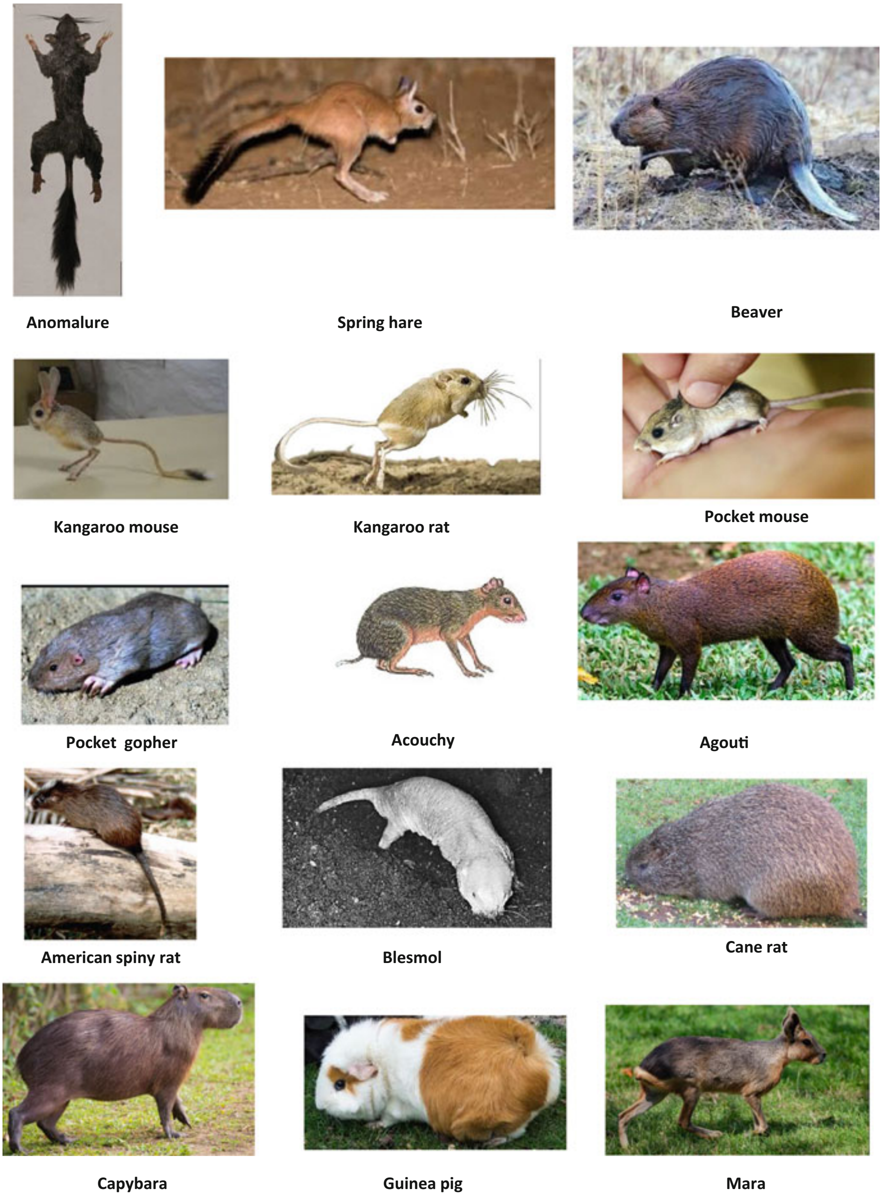
Members include amphibious and one of the largest rodents, viz. beaver (*Castor* sp.) and giant beaver (*Castoroides* sp., now extinct). They weigh about 16–30 kg. Beavers are native to North America, Europe and Asia. Beavers live in aquatic habitat and construct dams forming ponds that quite often cover many hectares. By this way, they alter the landscapes in which they live, and beavers are referred to as ‘ecosystem engineers’. Beaver incisors are strong and massive and have orange outer enamel containing iron in place of calcium. Beavers are colonial animals, and they construct dome-shaped island lodges with plant parts plastered with mud to protect the nest from predators.

11.3.3.2 Family Heteromyidae

Includes kangaroo mice, kangaroo rats and pocket mouse which falls under 36 families and 4 genera.

Kangaroo mouse (Plate 11.1) is a jumping rodent predominantly found only in the arid zones of the western parts of USA. Large head, large ears, fur-lined external cheek pouches, short forelimbs and long hindlimbs are the characteristic features of kangaroo mouse. The cheek pouches are used to carry food to the nest. Kangaroo mice produce concentrated urine and dry faeces as they do not drink water; instead, they obtain it from the food they eat.

Kangaroo rats (Plate 11.1), like the kangaroo mouse, have large heads and eyes, fur-line external pouches alongside the mouth, short forelimbs and very long hind



Anomalure

Spring hare

Beaver

Kangaroo mouse

Kangaroo rat

Pocket mouse

Pocket gopher

Acouchy

Agouti

American spiny rat

Blesmol

Cane rat

Capybara

Guinea pig

Mara

Plate 11.1 Rodent diversity

legs and feet. The tail has a characteristic tuft at the tip. Kangaroo rats can jump up to 2 metres (6.6 feet) using their hind legs. Like the kangaroo mice, kangaroo rats seldom drink water and get the same from their food. Kangaroo rats are night foragers and carry the food in their cheek pouches to the nest.

Pocket mouse (Plate 11.1) is an American rodent and has more than 36 species. Unlike kangaroo mouse, pocket mouse uses all four legs for the movement. Pocket mice consume many plant materials like seeds, nuts and succulent parts and carry the food in their cheek pouches.

11.3.3.3 Family Geomyidae

At least 38 species are present in this North and Central American rodent's family, and they are commonly called as pocket gopher. The 'pockets' are cheek pouches along the sides of mouth. Pocket gophers (Plate 11.1) are extensive burrowers that make shallow tunnels mainly using their forelimbs. One of the characteristic features of pocket gopher is that they can move backwards as fast as they move forwards. The pocket gophers feed mainly on plant roots and tubers.

11.3.4 Suborder Hystricomorpha

This suborder has 19 families.

11.3.4.1 Family Dasyproctidae: Includes Acouchy and Agouti

Acouchy is an antelope-like animal weighing about 1–1.5 kg (2.2–3.3 pounds), with a pencil-thin tail tufted at the tip (Plate 11.1), and is inhabitant of South America. Acouchys are terrestrial and nocturnal rodents with long, three-toed hind legs. When threatened, acouchys produce whistles similar to the ones produced by birds and hop a greater distance like the duiker, an African antelope. Acouchys eat seeds, nuts and other plant parts and also burry nuts in soil in their territories.

Agouti resembles a small forest-dwelling hoofed animal. It has an elongated body (Plate 11.1) and weighs up to 6 kg. The tail is very small and bald. The hindlimb toe has hoof-like claws. They are capable of jumping vertically up to two metres from a stationary position. Agoutis feed mainly on fruit, nuts and seeds; occasionally, they also feed on leaves, flowers, fungi and insects. Like acouchys, agoutis also bury nuts in the soil and disperse the seeds of many plants.

11.3.4.2 Family Echimyidae

American spiny rat (Plate 11.1) (18 genera, viz. *Proechimys,* terrestrial spiny rats; *Mesomys*, spiny tree rats; *Carterodon*, Owl's spiny rat; *Hoplomys*, armoured rat) is a nocturnal animal and has nearly 80 species. It is predominantly found in Central and South America. Like house lizards, the tail breaks off easily when pulled. They eat leaves and shoots of plants (bamboo in case of American bamboo rats), nuts, fruits, fungi and insects.

11.3.4.3 Family Bathyergidae

Blesmol is a burrowing naked mole rat with protruding incisors found in arid regions south of the Saharan deserts. Blesmols are colonial rodents living with nearly 300 individuals per colony. They appear to be without neck and external ears; however, they can sense the vibrations emanating from the ground. Underground roots, bulbs, tubers, other plant parts and occasionally invertebrates are the part of their diet. Blesmol is a prolific breeder and produces up to 27 pups per litre, which is the largest for any mammal.

11.3.4.4 Family Thryonomyidae

Cane rat, genus *Thryonomys*, is relatively larger rodent weighing up to 7 kg with blunt muzzles and small ears. Their fur coat is brown, rough and bristly (Plate 11.1). Cane rats are swift and agile on land and swim very well. They feed on grass, agricultural crops, bark of plants and fruits. Cane rats are hunted for human consumption in Africa.

11.3.4.5 Family Caviidae: It Includes Capybara, Guinea Pig and Mara

Capybara is also called as ‘water hog’, and it is the largest living and semiaquatic rodent of Central and South America (Plate 11.1). Largest capybaras are the South American species which grow about 1.25 m long and weigh up to 66 kg or even more, and the smallest one is the Panamanian capybaras (27 kg). Capybaras are generally brown coated and have almost no tail but with small ears, blunt snouts and short legs. They are vegetarian and at times become pest on cultivated areas.

Guinea pig is a domesticated rodent in South America. It has large head with large eyes and short ears; the body and the limbs are short (Plate 11.1). Several breeds of domesticated guinea pigs exist based on the coat texture and hair length. Coat coloration is extremely variable ranging from white to black. Domestic guinea pigs are fairly large, weighing between 500 and 1500 g. Domestication of guinea pigs started in Peru for more than 3000 years, and now there exists no wild population of this animal.

Mara resembles hares; it has a blunt muzzle, large ears and prominent eyes. The legs are slender and long (Plate 11.1). The body is 45–75 cm long, and the weight ranges between 9 and 16 kg. Their front claws are sharp but have hoof-like hind claws. Their diet is generally any available vegetation in the habitat.

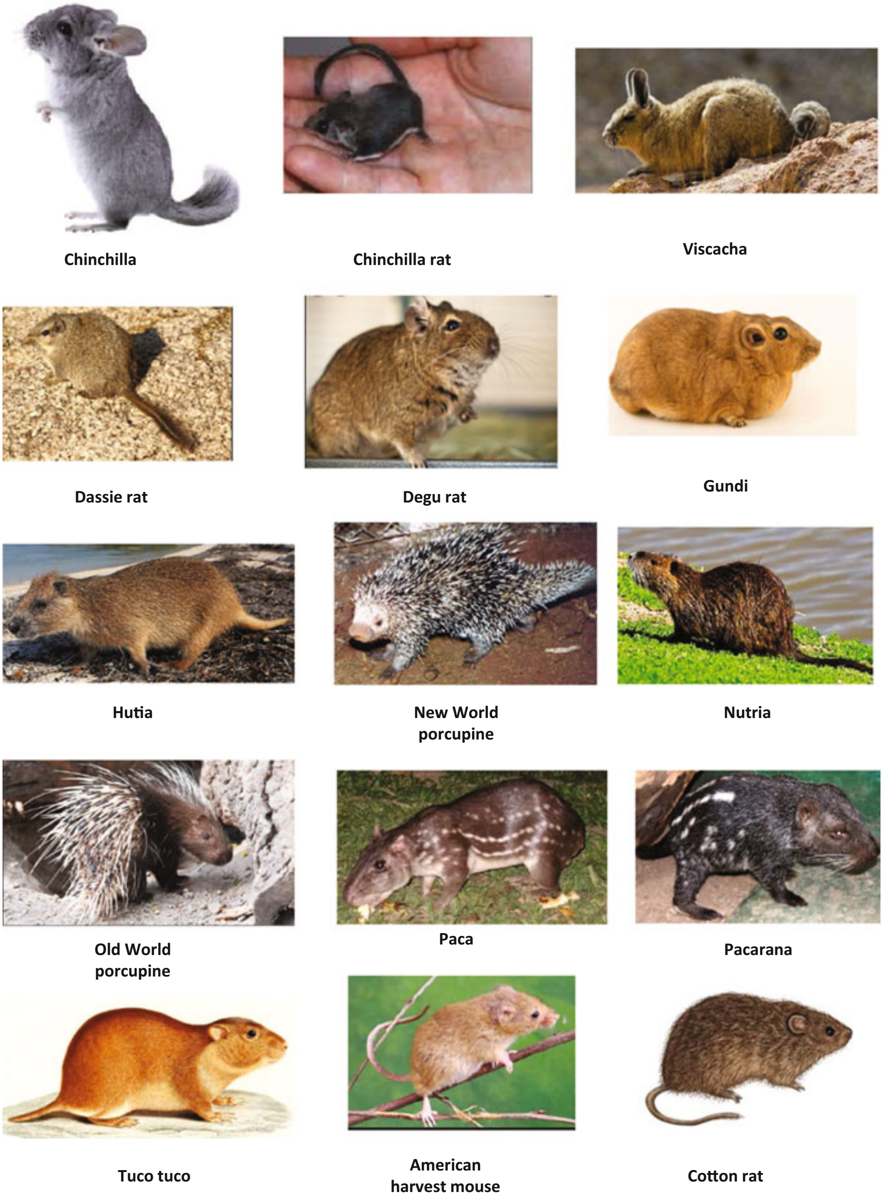
11.3.4.6 Family Chinchillidae Consists of Chinchilla and Viscacha

Chinchilla is a highly valued rodent for its soft and thick fur which is generally blue to brownish grey in colour (Plate 11.2). Chinchillas are hunted almost to extinction in the wild. However, they are commercially reared with various colours and kept as pets. Chinchilla has a compact body with large ears and eyes and bushy tail.

Viscacha resembles chinchillas and is an inhabitant of South America. They have dense soft fur. Their ears are very long and resemble long-tailed rabbits (Plate 11.2). They weigh up to 3 kg.

11.3.4.7 Family Abrocomidae: Chinchilla Rat

Chinchilla rat superficially resembles a chinchilla but is more rat-like with short limbs, large eyes and large, rounded ears (Plate 11.2). They are nocturnal animals and are agile rock climbers. Massive latrines are built by some species of chinchilla



Chinchilla

Chinchilla rat

Viscacha

Dassie rat

Degu rat

Gundi

Hutia

New World porcupine

Nutria

Old World porcupine

Paca

Pacarana

Tuco tuco

American harvest mouse

Cotton rat

Plate 11.2 Rodent diversity

rat on rock crevices using their faeces, urine and other liquids which eventually become rock solid. Chinchilla rats feed mainly on vegetation.

11.3.4.8 Family Petromuridae: Dassie Rat

Dassie rat is a rock dweller of southwestern Africa. It has a squirrel-like body (Plate 11.2) and weighs 170–300 g. Dassie rat can flatten its body against any substratum and can squeeze itself into very narrow gaps. They feed on grasses, fruits and other plant materials. Dassie rat is very unique among the rodents as it can regurgitate the food for further chewing more like the ruminants do.

11.3.4.9 Family Octodontidae: Degu (Genus *Octodon*)

Degu is a small rodent endemic to central Chile. It prefers to live in open grassy areas near human habitation. Their head and eyes are large sized, but they have medium-sized almost hairless ears and black-tipped tails (Plate 11.2); some animals have pale neck band also. Long, comb-like bristles project over claws on the hind feet, which is a characteristic feature of degu. Their body weight ranges between 170 and 300 g. Degus forage on the ground and also on shrubs and trees. Many individuals live together in a common burrow complex which has nests and stores.

11.3.4.10 Family Diatomyidae: Diatomyid

Fossil evidences are available for Diatomyidae from early Oligocene (~32.5 million years ago) to the Miocene (~11 million years ago). Recent addition to this group is *Inopinatia balkanica*, from the early Oligocene of southeast Serbia (Dawson et al. 2006; Marković, et al. 2018).

11.3.4.11 Family Heptaxodontidae: Giant Hutia

An extinct group of large rodents once lived in West Indies.

11.3.4.12 Family Ctenodactylidae: Gundi (Multiple Genera)

Gundis have two rows of comb-like bristles on their hindlimb for grooming themselves. Larger eyes, blunt nose and short but rounded ears are the identifying characters of gundi (Plate 11.2). Unlike other rodent species, gundis do not make burrows or nests; instead, they use available crevices and caves to rest at night. They eat leaves, seeds, flowers and other plant materials. When threatened, gundis produce birdlike sharp whistling alarm calls.

11.3.4.13 Family Capromyidae: Hutia (Multiple Genera)

Hutia is a Caribbean rodent with 26 living and recently extinct species. Hutias have stout body, short limbs, smaller eyes, exceptionally long whiskers and very prominent claws (Plate 11.2). They are specialised tree and rock climbers and dwellers, and like the gundis, they do not excavate burrows. Hutias are active both on day and at night. Some of the species like the long-tailed Cuban hutias resemble tree squirrels and are nocturnal. Plant materials like leaves, stem, bark, underground plant parts and sometimes small vertebrates constitute hutias' diet.

11.3.4.14 Family Erethizontidae: New World Porcupines

Members are medium-sized animals weighing up to 18 kg, and body is covered with modified hairs called spines or hollow quills. Some species are entirely arboreal and have long prehensile tails and wide foot pads; however, the less arboreal species have shorter tails. New World porcupines are solitary nocturnal animals and shelter themselves in tree caves, rock crevices or among the tree root networks (Plate 11.2). These rodents ingest nuts, leaves, bark, roots, grass, aquatic plants, insects and small reptiles. By the way of stripping the bark, these rodents kill the trees. Mineral requirement is met from the bones and antlers on which these rodents gnawed upon.

11.3.4.15 Family Myocastoridae: Nutria (*Myocastor coypus*)

Nutria, also called coypu, is originally from South America (southern and central parts). Fur of this animal has tremendous commercial value. Nutria is an amphibious rodent with large body (weight ranges between 10 and 17 kg). Hindlimbs with five toes and webbing are adapted to lead amphibious life, whereas the forelimbs have no webbing, but all toes have very well developed claws (Plate 11.2). Nutria can remain underwater for nearly 5 minutes. It can also close its mouth while cutting the submerged vegetation without swallowing water. In general, the nutria prefers slow-moving water and feeds on a wide variety of aquatic vegetations. Occasionally, they feed on aquatic invertebrates also. Nutria lives in families of 10 to 15 individuals, constructing burrows and platform nests in river banks. Intentional and accidental introduction of nutria in North America and Europe facilitated the nutria to become one of the destructive pests on cultivated crops.

11.3.4.16 Family Hystricidae: Old World Porcupine (genera *Atherurus*, *Hystrix* and *Trichys*)

Old World porcupines are large animals which move slowly. They are primarily terrestrial animals. Their primary defence is their imposing spines or quills (Plate 11.2); *Hystrix* sp. rattles its quills for communication among the individual and warn their predators. There are three groups of Old World porcupines, viz. a tree-climbing long-tailed porcupine (*Trichys fasciculata*), brush-tailed porcupine (*Atherurus* sp.) and short-tailed porcupine (*Hystrix* sp.). Among the three, *Hystrix* sp. is the largest (nearly 30 kg body weight). They feed on many kinds of plant material and also carrion. They gnaw antlers and bones to get calcium and other minerals. Even though they are excellent diggers, they often shelter in caves and rock crevices.

11.3.4.17 Family Cuniculidae: Paca (Genus *Cuniculus*)

Paca is a terrestrial herbivorous rodent in South America. It has pig- or deer-like body which weighs between 6 and 14 kg, square head and a tiny tail (Plate 11.2). Pacas reside in rain forests and near water bodies. They are excellent swimmers and can stay underwater up to 15 minutes, at times. Pacas live in deep burrows (3 m) near water bodies. These rodents make peculiar growling sound using their cheeks. Pacas diet consists of fruits, leaves, fungi and insects; they are also coprophagous and absorb nutrients from fresh excreta.

11.3.4.18 Family Dinomyidae: Pacarana (*Dinomys branickii*)

Pacarana is a rare rodent found in tropical rain forests of Amazon river basin and Andes mountains in South America. It has a robust body (weighing 10–15 kg), large head and eyes and unusually long vibrissae (Plate 11.2). Pacarana produce a variety of vocalisations for communication apart from stamping its fore paw. While eating, pacanas hold their food in their forelimbs and while sitting on their hindlimbs.

11.3.4.19 Family Ctenomyidae: Tuco-Tuco (Genus *Ctenomys*)

Tuco-tuco is a fossorial rodent similar to pocket gopher (Plate 11.2), of central and southern South American origin. Tuco-tuco makes a characteristic alarm calls, hence the name. These rodents spend most of their lives underground. Their forelegs are exceptionally long and equipped with powerful claws suited for digging; they also use their tooth in excavation process. Tuco-tucos lead either solitary or semi-social lives feeding on vegetation, root, bark (considered as agricultural pest), invertebrates, small mammals, reptiles, toads and even birds.

11.3.5 Suborder Myomorpha

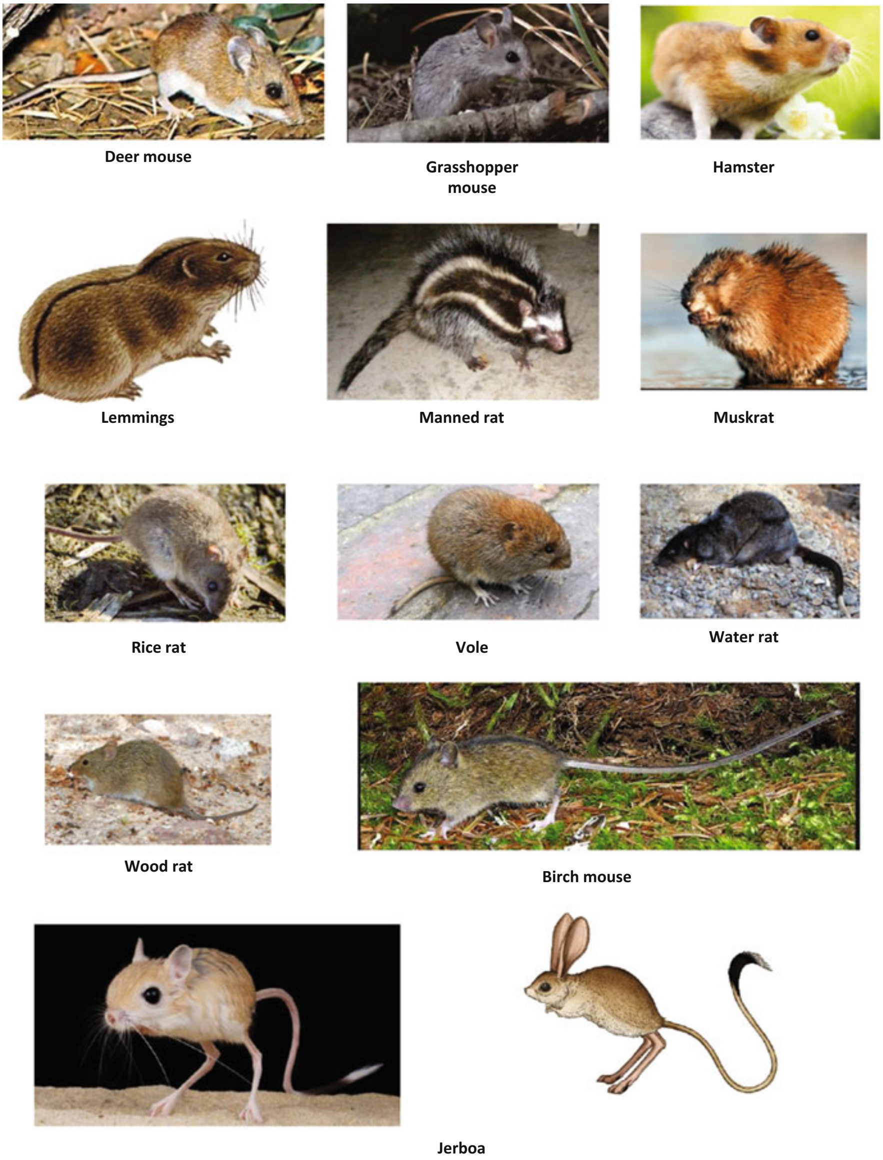
11.3.5.1 Family Cricetidae

It is the second largest mammalian family having nearly 608 species; it includes New World rats and mice, hamsters, lemmings and voles.

American harvest mouse: There are 20 species of American harvest mice found in southern Canada, USA, Mexico to northern South America. An adult harvest mouse barely weighs around 20 g (Plate 11.2). Harvest mice are nocturnal and use the runways of other rodents. At low temperature, these animals enter into torpor. They are herbivorous animals and also pests of forest trees and cultivated crops. Their diet consists of grains and seeds of various plants.

Cotton rat (genus *Sigmodon*) is found in southern and northern parts of USA and South America, respectively. They have S-shaped molar, hence the genus name, *Sigmodon* (literally means S-tooth). All species live in natural grassland habitats, and all are primarily herbivorous and inhabit cultivated fields. They are both nocturnal and diurnal in habit (Plate 11.2). Cotton rats lead a solitary life except during mating season. They eat seed and plant materials; thus, they can become serious agricultural pests.

Deer mouse (genus *Peromyscus*) is small rodent with conspicuous bulging eyes found in North and Central Americas. Deer mouse genus, *Peromyscus*, has 56 sub-species, and the body weight of different species ranges from 15 to 110 g. Fur is soft and colour varies from nearly white to grey, brown, reddish brown, to black, white underside (Plate 11.3), and all have white foot. The white-footed mice are nocturnal, and their reproduction depends on the availability of food. They are one of the preferred laboratory animals for the study on genetics, evolution, physiology and medicine. Incidentally, *P. maniculatus* is carrier of dreaded Hanta virus, plague and Lyme disease.



Deer mouse

Grasshopper mouse

Hamster

Lemmings

Manned rat

Muskrat

Rice rat

Vole

Water rat

Wood rat

Birch mouse

Jerboa

Plate 11.3 Rodent diversity

Grasshopper mouse (genus *Onychomys*) is a terrestrial, nocturnal, insectivorous and carnivorous rodent (Plate 11.3) adapted to semiarid and arid habitats; they are endemic to North America. Grasshopper mice mostly eat grasshoppers, other insects and arthropods like scorpions; they are also known for stalk and kill small rodents and snakes and eat them just like the larger mammalian predators. They use a variety of vocalisation to communicate, and the calls are used to distinguish species, sex and even the position in the hierarchy (immature and adults make different vocalisation); the howling calls these rodents make, like the miniature version of a coyote howl, can be heard over long distances.

Hamsters are popular house pets in many countries (e.g. golden hamster or Syrian hamster, *Mesocricetus auratus*) (Plate 11.3). Hamsters have silky fur, and the colour ranges from black to red depending on the species. They are solitary animals, and if put together, the animals show acute and chronic stress. Hamsters are omnivorous animals and consume seeds, grassy vegetation, invertebrates and even other small animals. Hamsters are known to carry food in their cheek pouches to their burrow.

Lemmings are small rodents and found only in the Northern Hemisphere. There are 20 species, some of which undertake large, swarming migrations. They have soft fur-covered rounded body with short legs and stumpy tails (Plate 11.3). Among the different species of lemmings, the wood lemming (*Myopus schisticolor*) and steppe lemming (*Lagurus lagurus*) are the smallest, weighing about 20 to 30 g. Lemmings are behaviourally different from other rodents as they aggressively behave towards predators. Some species of lemmings undertake migrations, and if the migration is not successful through a large water bodies, many of the migrating lemmings die *en route*, which leads to the popular notion, ‘suicidal lemmings’. They feed on almost any sort of vegetation like grass and mosses. Lemmings make extensive runway systems under the rock or snow and scamper along.

Maned rat (*Lophiomys imhausi*) is also called crested rat which resembles a porcupine and found in East Africa. Mane is the coarse, black and white banded hairs present on the head and extends beyond the tail base; when alarmed or excited, the mane gets erected (Plate 11.3)**.** These rats are known to deliberately smear poisonous materials from plants on which they chew as a defence mechanism. The maned rat sits on its haunches when eating and manipulates food with its forelimbs. They feed on fruits, leaves and other plant materials; they also consume insect or meat, if available.

Muskrats (genera *Neofiber* and *Ondatra*) are large amphibious rodent; they are native to North America but found also in Europe and Asia. These rodents produce a musky substance from their perineal glands to mark their home ranges. These rodents also are called as musk beavers because of the flattened tail. Muskrats use their large semi-webbed hind feet as oars while swimming (Plate 11.3). They are very efficient swimmers (5 km/h) and divers (can stay submerged for up to 20 minutes). Muskrats consume various aquatic plants; occasionally, they also feed on aquatic animals and young birds. Muskrats usually live in groups and build nests to protect themselves and their offspring. These rodents are being hunted for their fur and meat.

Rice rats (genus *Oryzomys*) including about 120 species are small rodents (Plate 11.3) found in many environments, viz. coastal marshes, grasslands and rainforest of USA and South America. They resemble house rat but are smaller in size. An opportunistic feeder, they eat a variety of food materials like plant seeds, succulent parts of grasses, invertebrates, small vertebrates and even carrion.

Voles are also called as meadow mice. They resemble shrews, rats, mice and gophers; they are small-bodied rodents with stouter body and shorter but hairy tail found in the Northern Hemisphere. Voles live in a wide variety of habitats eating plants, nuts, fruits, dead animals, etc. Voles are known to griddle young trees which eventually kill the plant; they also eat roots until the plant is dead. However, the voles play a vital role through their burrowing activities to disperse soil nutrients in different soil layers.

The **woodland vole** (*Microtus pinetorum*) is one of the smallest voles weighing less than 35 g. It has brown dorsum and silvery underside. It is found in the apple orchards, dry fields and deciduous forests of eastern USA. Woodland vole inflicts heavy damage to apple orchards.

The **meadow vole** (*Microtus pennsylvanicus*), also known as the meadow mouse, has the widest distribution in North America. Meadow voles prefer grasslands, but they are found in forest areas also. These rodents dig shallow burrow, and nest is constructed with woven grass. Diet includes agriculturally important plant species, grasses and sedges; the meadow voles take insects and snails and, at times, carrion feeder.

Water rats are a group of not closely related semiaquatic carnivorous rodents (Plate 11.3). They live in burrows dug near the water bodies. They are expert swimmers. The animals feed on a variety of aquatic insects, crustaceans and small fishes. Their sensitive whiskers aid in locating the prey underwater.

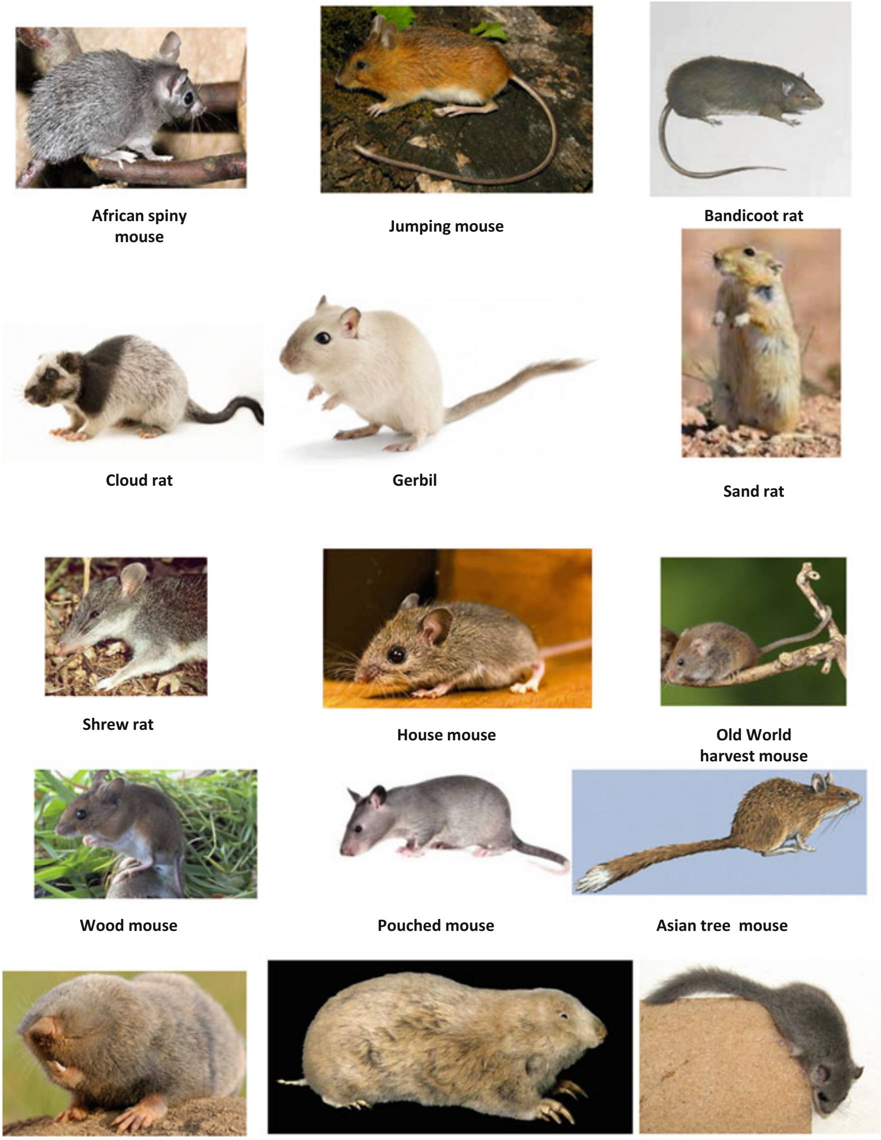
Woodrats (genus *Neotoma*), also called packrats, are medium-sized North and Central American rodents (Plate 11.3). Middens (collection of various items includes bones, sticks, dry manure, shiny metal objects, etc.) are seen near their dwellings. Woodrats are generally solitary, nocturnal and active year-round. Normally, wood rats feed on green vegetations and some species, viz. Stephen's woodrat (*Neotoma stephensi*) thrive on juniper sprigs and *N. albigula* and *N. lepida* on cacti and yucca plants.

11.3.5.2 Family Dipodidae: Dipodid/Birch Mouse (Genus *Sicista*)

Birch mice are small, long-tailed jumping mouse-like rodents (Plate 11.3) found in Europe and Asia. They are nocturnal in general, but also seen in shallow burrows during daytime. They feed on seeds and insects and live in nests constructed out of woven ball of grass either in bush or crevices. Birch mice move around by leaping on the ground, but they use their tail for support while climbing on the tree.

Jerboas are hopping rodents of Europe, Asia and northern Africa found in grasslands and deserts. Jerboas resembles mouse but have short forelegs and extremely long hind legs for jumping; when alarmed, these rodents can leap up to 3 m. The tail is often tufted (Plate 11.3). Jerboa's dense fur colour usually matches the ground of the animal's habitat. In some species of jerboa, to regulate the temperature and moisture in their burrows, plugging the burrow entrances with soil is noticed; it also prevents the hot air getting into their burrow network.

Jumping mouse (genera *Eozapus*, *Napaeozapus* and *Zapus*) is a small (13–26 g), mouse-like rodent found in North America and China. These rodents have elongated hind legs for leaping when alarmed (4 m high) and have tail which is longer than the head and body (Plates 11.4) which is used as a balancing organ. They have soft and



African spiny mouse
Jumping mouse
Bandicoot rat
Cloud rat
Gerbil
Sand rat
Shrew rat
House mouse
Old World harvest mouse
Wood mouse
Pouched mouse
Asian tree mouse
Blind rat
Zokor
Dormouse

Plate 11.4 Rodent diversity

glossy fur, and the fur colour varies. Jumping mice are generally terrestrial but are agile climbers and swimmers. They feed on seeds, fruits, fungi, invertebrates and sometimes molluscs and small fishes, also.

11.3.5.3 Family Calomyscidae

Mouse-like hamster (living fossils) is a group of small rodents, found in rocky areas in desert regions of Central Asia.

11.3.5.4 Family Muridae

African spiny mice (genus *Acomys*), which originated from Africa, have unusually stiff hairs similar to spines on their coats, hence the common name. They have large eyes and ears and scaly tails (Plate 11.4). African spiny mice are social animals and are omnivorous; they eat plant materials like dates and also some odd diet like dried flesh and bone marrow of mummies in Egypt. They live in a variety of environment like rocky areas, savannas and dry woodlands; they occupy rock crevices, termite mounds or even other rodent burrows. Skin of these mice is brittle and can be easily torn (an escape mechanism from predators), which can be completely regenerated, a special trait that no other mammals possess.

Bandicoot Rat (Genera *Bandicota* and *Nesokia*)

Bandicoot rat is a commensal rodent, and there are five Asiatic species which depends on human habitations for their survival. They are found throughout the Indian subcontinent and Indochina. The larger bandicoot rat (*Bandicota indica*) that weighs up to 1 kg (Plate 11.4) is a robust blackish brown rat with a sparsely haired tail equal to the head-body length. These rats erect their hairs on their back and produce grunts when challenged. They cause extensive damage to masonry structures with their burrowing activities, and they are also very important carrier of many zoonotic diseases. These rats consume any household refuse, grains and vegetables and are serious pest on poultry.

The lesser bandicoot rats (*B. bengalensis*) or the Indian mole rats are one of the large rats in Southern Asia. They have dark brown or brownish grey body fur and weigh up to 350 g and measure up to 40 cm long including their brown tails which is shorter than the head-body length. The lesser bandicoot rat is found on the Sothern and Southeast Asia, Middle East and Africa. They produce pig-like grunts when threatened. They are extensive burrowers, and the elaborate burrows have numerous chambers and exit or bolt holes. They are very serious pest on agricultural crops; they store grains and other food materials in their nest. These rodents can burrow through even concrete cellars.

Savile's (*B. savilei*) bandicoot rats are solitary omnivorous animals found in the Indo-Malayan Realm (Myanmar, Thailand and Vietnam). They resemble more like the lesser bandicoot rat. They feed on grains, fruits and invertebrates and a serious pest on agricultural crops.

Short-tailed bandicoot rats (*Nesokia*) are of the size of the bandicoot rats with soft brown fur on the upper parts, lighter underpart and a short tail with scantily haired. They live in river valleys and irrigated crop lands in Asia and Africa. The rats make extensive burrow system which goes up to 60 cm deep and 9 m long; they push up mounds of soil to conceal the entrance and exit holes. Some species of *Nesokia* are excellent swimmer and live in natural marshes and build nests on reed platforms above water level. The diet of these rats includes grains, grasses, roots, fruits and vegetables.

Cloud rats (genera *Phloeomys* and *Crateromys*), also called cloud runner, are large, slow-moving endemic arboreal rodents of Philippine forests (Plate 11.4). They are nocturnal and herbivorous. Cloud rats have been hunted for their meat driving them to near extinction.

Gerbils (subfamily Gerbillinae), once known as desert rodents, are living in arid habitats and have more than 110 species. They are social animals, and a gentle and hardy gerbil, *Meriones unguiculatus*, is even kept as a popular house pet (Plate 11.4) in some regions. Most gerbils are nocturnal. Their hind legs are long, and they have large ears and eyes. When alarmed, the gerbils flee in running leaps. They are known to construct elaborate underground galleries, and the burrows of the great gerbil sometimes weaken embankments. In the wild, gerbils inhabit often harsh environments with little vegetations. They consume plant materials and insects and occasionally, like the Indian gerbil (*Tatera indica*), eat eggs and young birds.

Mouse (genus *Mus*) is a small rodent (about 5 inches long) having 38 species found worldwide (Plate 11.4). The subgenus includes spiny mice (*Pyromys*), shrew-mice (*Coelomys*), rice field and the house mice (*Mus*) and African mice (*Nannomys*). The common house mouse (*Mus musculus*) is native to Central Asia, and now it is a global commensal rodent.

Mice have a slender body, pointed snout, round small ears and scally tail which is equal to the head-body length. Fur colour varies among the species of *Mus.* Body weight of pygmy mouse (*Mus minutoides*) of sub-Saharan Africa is only 3 g, whereas the largest flat-haired mouse (*Mus platythrix*) of peninsular India weighs up to 18 g. Mice are nocturnal rodents and a few are specialised burrowers which live in grasslands. Basically herbivorous but mice adapt very well to human habitation and feed nearly anything digestible. Mice are prolific breeders producing up to 14 litters per year (1 to 12 offspring per litter).

Old World Harvest Mouse (Genus *Micromys*)

Old World harvest mouse (*Micromys minutus*) is the smallest of rodents (weighing less than 7 g) with semi-prehensile tail and lives in the Northern Hemisphere (Plate 11.4). These nocturnal rodents are good climbers and prefer to live among tall vegetation. They construct globular nests of grass suspended between branches and stems. These mice eat seeds and other vegetation in addition to insects and the eggs of small birds.

Old World Rat (Genus Rattus)

Rats (genus *Rattus*) or true rats originally belong to continental Asia. Brown rat, *Rattus norvegicus*, and the house rat, *Rattus rattus*, however, spread far and wide. These commensal rodents eat human food resources, damaging and contaminating stored grains and killing domestic poultry. They created havoc in island ecosystem driving endemic fauna to their extinction. Numerous zoonotic diseases have been spread by these rodents. However, the brown rat has been used in laboratories worldwide for medical, genetic and biological researches.

Rats in general have a slender body with pointed head, large eyes and prominent ears. The brown rat is bigger than the house rat, and its tail is shorter relative to the body length. The brown rat also has thicker furs and 12 pairs of mammae instead of 10 compared to the black rat.

Being a commensal species, the house rat consumes nearly anything digestible with particular preference to grain and fruits. However, the brown rat is basically omnivorous but prefers a carnivorous diet and even resorts to carrion feeding. Other tropical species, such as the rice-field rat (*R. argentiventer*) and Malayan field rat (*R. tiomanicus*), primarily consume the insects, snails, slugs and other invertebrates found in the habitats. Rats are prolific breeders, start reproducing within three months and produce up to 12 litters of 2 to 22 pups (8 or 9 is usual) per year.

Shrew rats (various genera, viz. *Chrotomys, Echiothrix, Rhynchomys, Archboldomys, Microhydromys, Pseudohydromys, Celaenomys, Mayermys, Melasmothrix, Neohydromys*) are carnivorous ground-dwelling rodents. They are endemic to the tropical islands of Philippines and New Guinea.

These shrew-like rodents have small eyes and long whiskers (Plate 11.4). The Sulawesi spiny rat is the largest shrew rat (weighs 220 to 310 g). Shrew rats of New Guinea are very small (*Microhydromys richardsoni*) and weigh only 9 to 12 g. Shrew rats always poke their noses through wet leaf litter and moss to locate their food, mostly the earthworms.

Water rats (genera *Hydromys*, *Crossomys* and *Colomys*) are amphibious carnivorous rodents (Plate 11.4). They swim with their long hind legs equipped with webbed digits and hunt food underwater with the help of highly sensitive whiskers.

Wood mice (genus *Apodemus*) are small-bodied rodents, native to Europe and northwestern Africa. Body weight of these nocturnal rodents ranges between 15 and 50 g and body length between 6 and 15 cm excluding the tail (Plate 11.4). If caught by tail, these rodents shed the tail which never regrows. Wood mice live in burrows and live in forests, grasslands and cultivated fields. They feed on seeds of oak and other trees, roots, fruits, insects, snails and hibernating bats.

11.3.5.5 Family Nesomyidae: Nesomyid (Genera *Beamys, Cricetomys* and *Saccostomus*)

African pouched rats (subfamily Cricetomyinae) have characteristic cheek pouches. All terrestrial nocturnal animals have grey to brown coats with white or grey under parts (Plate 11.4). These are the largest muroids (about 3 feet long including the tail) in the world. These are gentle animals and can easily be tamed and raised in captivity. They are nocturnal, colonial and omnivorous animals found throughout sub-Saharan Africa. They become invasive in northern America. Gambian pouched rats apparently have the calibre to detect land mine and tuberculosis.

The short-tailed pouched rats (genus *Saccostomus*) are small rodents and inhabit savannahs, steppe and cultivated lands in Africa. Their legs are short and adapted for digging to make their own burrows, but they also use cavities made by other animals, holes among tree roots and rock piles to nest. These rats usually inhabit natural grasslands and cultivated land. They primarily feed on seeds during wet periods but eat insects during drought. A characteristic feature of these rodents is that they go spontaneously into torpor with sudden variation in temperature.

The long-tailed pouched rat (*Beamys hindei*), also called as lesser hamster rat, is nocturnal and a nimble climber. They have medium-sized body (weighs up to 97 g) and have a scantily haired tail about as long as the head and body. It constructs burrows in soft sandy soil. This species lives in forest and open woodlands and is also found in fallow agricultural fields and feeds mainly on fruits and seeds.

11.3.5.6 Family Platacanthomyidae: Oriental Dormouse

Asian Tree Mouse (Genera *Platacanthomys* and *Typhlomys*)

Asian tree mice (genera *Platacanthomys* and *Typhlomys*) are small rodents found only in a few tropical forests of India and continental Southeast Asia.

The Malabar spiny tree mice (*Platacanthomys lasiurus*) are endemic to Western Ghats of India. Though they resemble dormouse, both are not closely related. The ears of the rodent are pointed at the tip. They are nocturnal, frugivorous and arboreal; they build nests in tree cavities and eat fruits and nuts. The hairs on the tail are long and white at the tip of the tail resembling a bottlebrush (Plate 11.4).

11.3.5.7 Family Spalacidae: Spalacid (Genera *Rhizomys* and *Cannomys*)

Bamboo rats are burrowing, slow-moving, nocturnal rodents endemic to Asia. Bamboo rats have a robust, cylindrical body, small ears and eyes and short stout legs (Plate 11.4).

Bamboo rats construct a simple burrow with single entrance and exit holes, a nest and a latrine chamber. To dig they use their incisor teeth and claws. *Rhizomys* species construct their burrows among roots of bamboo stands and line their nest with dried grass and shreds of bamboo. They feed on bamboo roots and also roots of sugarcane and cassava.

The lesser bamboo rat is a mole-like burrowing animal digs deep tunnels in the rocky ground of mountain area, forest floors and even gardens; mount of excavated soil is found at the entrance of the burrow hole. It feeds mainly on bamboo shoots, roots and other vegetation; it is also found in tea plantations.

Blind mole rats (genera *Nannospalax* and *Spalax*) are fossorial blind rats. Their small eyes are completely covered by a layer of skin. They have cylindrical body, short limbs and protruding incisor teeth (Plate 11.4) which is used for digging purpose; they possess a minute stub of a tail which may not be visible externally. Their diet includes roots, tubers and bulbs which they search 10–25 cm below-ground, but occasionally they feed on aboveground seeds and green plant parts.

Zokors (genus *Myospalax*) are Asiatic fossorial rodents resembling mole rats with cylindrical bodies with short powerful limbs. They have long self-sharpening front claw. The tiny eyes are very sensitive to light and nearly hidden in furs (Plate 11.4)**.** Zokors are efficient burrowers, and the main burrow is dug about 2 metres below the surface and is constructed with separate chambers for nesting, food storage and waste. They feed on roots, bulbs and rhizomes and occasionally leaves and shoots.

11.3.6 Suborder Sciuromorpha

11.3.6.1 Family Gliridae: Dormouse (Various Genera, viz. *Graphiurus, Dryomys, Myomimus, Eliomys, Selevinia, Glirulus, Muscardinus and Glis*)

Dormouse (family Myoxidae) is a small-bodied nocturnal rodent found in Africa, Asia and Europe. Dormice have large eyes, rounded ears, short legs and hairy or bushy tails (Plate 11.4). They are known for their prolonged hibernation in winter. They have soft and dense reddish brown fur; dark stripe along the back and dark facial markings are seen on some species.

Dormice construct globular nests and also use abandoned nests of other animals. They feed on fruits, nuts, invertebrate like insects and spiders, bird eggs and nestlings and small rodents; they are even cannibalistic.

Desert dormice (*Selevinia betpakdalaensis*) are a rare nocturnal rodent of Central Asia. They live among the desert vegetations on which they climb efficiently. They are endangered rodents living in clay and sandy deserts in southern Kazakhstan.

11.3.6.2 Family Aplodontiidae: Mountain Beaver

Mountain beaver (*Aplodontia rufa*) is a North American rodent. Extremely short tail and smaller body size (body length less than 50 cm and body weight less than 2 kg) differentiates from the American and Eurasian beavers (genus *Castor*). Small rounded ears with white spots, small eyes and short limbs with five digits are the identifying characters (Plate 11.5) of mountain beaver. Mountain beavers are asocial animals having very acute tactile and olfactory senses but limited auditory function and vision. The rodents move very little from their burrows, and the burrow entrance is covered with wilted vegetations. Diet includes succulent vegetation and fern.

11.3.6.3 Family Sciuridae: Squirrel

Family Sciuridae has 50 genera and 268 species. Squirrels are small- to medium-sized rodents which have bushy tails. Closely related to dormouse and mountain beaver, squirrels, viz. ground squirrels, chipmunks, marmots, prairie dogs and flying squirrels, occupy a range of ecological niches worldwide.

Tree squirrels have slender bodies and muscular limbs. The bushy tail is equal or even longer than the head and body length. Large, strong, curved and very sharp claws enable the tree squirrels to traverse among the branches of trees with ease. Tree squirrels consume varied foods like nuts, berries, fruits, flowers, bark, plant sap, insects and other invertebrates, eggs of birds, baby birds, etc. Sizes of tree squirrels vary; Oriental giant squirrels (genus *Ratufa*) weigh about 1.5 to 3 kg, whereas the pygmy squirrels, the smallest being the African pygmy squirrel (*Myosciurus pumilio*) of the West African, weigh only 13 to 20 g. Fur colour varies in tropical species from white to black.

Chipmunks (genus *Tamias*) are small, striped, terrestrial red squirrels, which are active during day (Plate 11.5). Their body length ranges from 8 to 16 cm and tail length almost equal to the body length. Their eyes, ears and tail are prominent, but the claws are delicate in contrast to the tree squirrels. Chipmunks are well adapted to

Mountain beaver

Chipmunk

Flying squirrel

Ground squirrel

Suslik

Marmot

Ground hog

Prairie dog

Tree squirrel

Plate 11.5 Rodent diversity

exploit the resources of rocky terrain and forest under stories. They inhabit various forest types and make characteristic chipping calls. They are omnivorous animals that eat grass shoots, seeds, fruits, nuts, berries, flowers and fleshy plant parts, fungi, arthropods and sometimes carrion feeding. They store nuts and seeds for future use in their nest.

Flying squirrels (multiple genera) are the gliding squirrels (Plate 11.5). They glide by extending their limbs supported by flaps of skin up to 450 m (Giant flying squirrels *Petaurista sp.*). These are nocturnal squirrels that live in tree cavities or globular nests they make with leaves, shredded bark or mosses on high trees. They feed on plant materials like seeds, fruits, leaves, flower buds, nuts, pollen, ferns, plant sap, lichen, fungi, arthropods, small birds, eggs, snakes and even smaller mammals.

Ground squirrels are ground-dwelling diurnal rodents. They are gregarious squirrels and live in colonies in open areas and grasslands. They have short limbs with strong claws and moderately long tail. Coat colour varies widely among the species (Plate 11.5) which may be solid or pattern of colours. Most ground squirrels are omnivorous and feed on fruits, nuts, fungi and seeds; they eat insects, other arthropods, eggs, birds and even other rodents.

Tropical Ground Squirrels

Tropical ground squirrels are small social rodents and active year-round. These squirrels do not store food. Tropical ground squirrels are paler and have unringed tail. There are five genera (*Dremomys*, *Lariscus*, *Menetes*, *Rhinosciurus* and *Hyosciurus*) under this group, and they live in the forests of Southeast Asia. These squirrels eat more arthropods than that of nontropical ground squirrels. During the period with scarce food availability, these squirrels undergo aestivation for a brief period.

Marmots (genus *Marmota*) are relatively large ground squirrels weighing 3 to 7 kg, depending on the species. They are found in North America, Europe and Asia. Marmots are brown and have short but robust legs and strong claws for digging. Their coat colour may be brown or black or a mixture of grey and white (Plate 11.5). They eat grasses, berries, flower, roots of plants, lichens and mosses.

Prairie dogs (genus *Cynomys*) are herbaceous burrowing rodents of North America that closely resemble marmots (Plate 11.5). Prairie dogs are stout bodied with a short tail and weigh approximately 1.5 kg. Prairie dogs dig elaborate burrow systems with volcano-shaped entrances and live in colonies called as ‘towns’. The ‘towns’ may cover hundreds of hectares, the largest ever recorded being in Texas that stretches across 65,000 square km and contained whopping 400 million individuals. They forage on succulent vegetation, seeds and roots depending on the season.

11.4 Behaviour of Rodents

Depending on the family and species, rodents occupy a variety of habitats and exhibit a wide range of behaviour, habits and lifestyle (Buckle and Smith 2015). The rodents may be burrow formers (gophers, rats and moles), or they may be living entirely on trees (flying squirrels) or leading aquatic life and spend most of their time in water (capybara) or living in some of the challenging terrain like deserts (kangaroo rats and jerboas). Generally, many rodents lead a social life, living in large groups with a complex social setup and interacting with each other frequently; prairie dogs, naked mole rats and ground squirrels live in large colonies. There are many other rodent species that live in smaller groups, like the beaver, containing the adult male, female and their offsprings. Specific territories exist for each colonies, and breaching the territory leads to violent fighting. Within the territory, there are numerous animals, and the colonies are broken into smaller units. For example, the prairie dog towns are divided into certain neighbourhoods. Within these smaller units, they look after the young ones; cooperate building other nests, playing together, mutual grooming and communication among the animals take place. In contrast to the social living, some rodents are solitary (many desert species, porcupines, pocket gophers and pocket mice), live lonely and feed alone; only during breeding time, these animals are seen with the opposite sexes for a brief time. Some of these solitary species construct and live alone in their own burrow system except during certain periods; more than one individual in the burrow during the mating season or the female animals with her dependent offsprings may live together for a brief period.

Except a few cases, most of the rodents are active throughout the year. At very low temperature in the temperate regions, some rodent species (ground squirrels) may suspend the activities and hibernate for several months or aestivate for a brief period when there is a scarcity in food availability, as in tropical ground squirrels. Elaborate communication system exists among rodent species; they communicate with numerous sounds, characteristic odours and sights. The innate capacity to reproduce is one of the primary reasons for the success of rodents, and they produce large numbers of offspring in one go. The strategy is having a short gestation period (17 to 45 days), having multiple litters per year and thus producing large numbers (can be up to or more than 20) of highly dependent babies (e.g. mice, rats and pocket gophers). Another strategy is rodent having longer gestation periods (60 to 238 days), fewer litters per year (generally one to two) and having a relatively fewer number of offspring with reasonably good parental care. Depending on the species, the mating system varies in rodents; there are monogamous rodents where the male and female pair is together for multiple mating seasons seen as in Patagonian mara. Harem-based mating system exists in other species where one male mates with a set group of females for the mating season. However, many rodents are promiscuous, meaning they mate randomly. As rodents are prolific breeders, availability of food determines the copulation built up in crop fields and storehouses.

Table 11.1 Rodent species of economic importance

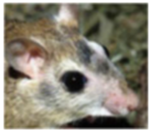
S. No	Family	Common name	Name of species
1	**Sciuridae**	Five stripped/Northern palm squirrel	*Funambulus pennantii* Wroughton
2		Three striped/Southern Indian palm squirrel	*Funambulus palmarum Linn.*
3	**Hystricidae**	Indian crested porcupine	*Hystrix indica* Kerr
	Muridae	**S.F. Gerbillinae**	
4		Indian desert gerbil	*Meriones hurrianae* Jerdon
5		Indian gerbil	*Tatera indica* Hardwicki
6		Hairy footed gerbil	*Gerbillus gleadowi* Murray
		S.F. Murinae	
7		House/black/ship rat	*Rattus rattus (Linn.)*
8		Soft furred field rat	*Millardia meltada (Grey)*
9		Norway rat	*Rattus norvegicus Berkenhout*
		S.F. Murinae	
10		House mouse	*Mus musculus* Linn.
11		Brown spiny mouse	*Mus platythrix* Bennett
12		Indian field mouse	*Mus booduga* Grey
13		Short-tailed mole rat	*Nesokia indica* (Grey and Hardwicke)
14		Lesser bandicoot/mole rat	*Bandicota bengalensis* (Grey)
15		Larger bandicoot/mole rat	*Bandicota indica* (Grey)

Rodents may be neutral or beneficial or harmful. As pest, rodents cause substantial damage and economic losses to human interests. Rodent pests may be in the field (field rats) or in the human habitation (commensal rats) (Tables 11.1, 11.2 and 11.3). Since they are prolific breeders, lots of efforts have been put forth to manage the rodent pests. Hereafter, mostly pest rodents are dealt with, unless or otherwise mentioned for management point of view.

11.4.1 Neophobia and Bait Shyness

Neophobia (neo = new, phobia = fear) is the fear of new things in rodents, has been a major issue encountered for at least four centuries in poison baiting of problematic rodents. The neophobic response can be one of the most important obstacles in the successful rodent management. Barnett (1958) defined neophobia as, 'the avoidance of an unfamiliar object in a familiar place'. The success of the rodent management programme depends on this behaviour of the rodents as neophobic rodents tend to avoid new foods and even foods previously eaten if they are placed on or in new objects or containers. The neophobic response varies between species, populations of the same species and even between individual animals. Female brown rats are considered to be highly neophobic which makes them difficult to trap or to attract

Table 11.2 Comparison of characters of selected field rats

Field rodents					
Species name	Body	Muzzle	Ear pinna	Tail	Faecal pellet
1. Lesser bandicoot rat and Indian bandicoot	 Heavy and thick body	 Blunt	 Small ear pinna, half buried in body	 Short tail; shorter than head-body (HB)	Bulky oval
Field mouse, *Mus booduga*	 Small, slender	 Small, pointed	 Small oval	 Shorter than HB, thin and naked	Qty: 50 to 75 pellets daily; size: ¼ inch long; shape: small with one or both ends pointed
Indian gerbil, *Tatera indica*	 Medium-sized body with big eye	 Pointed	 Large, prominent and stands out from the body	 Tuft of hairs at the tip	Oval at both ends

(continued)

Table 11.2 (continued)

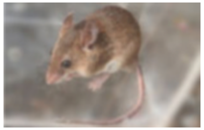
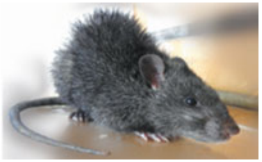
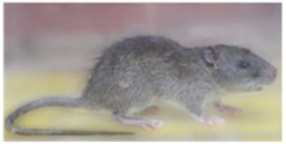
Field rodents					
Species name	Body	Muzzle	Ear pinna	Tail	Faecal pellet
Brown spiny mouse, *Mus platythrix*	Small and slender body	Small, flat and blunt	Large, fan shaped and stands out from the body	Naked, shorter than HB	Small elliptical

Table 11.3 Comparison of characters of commensal rats

Commensal rodents					
Species name	Body	Muzzle	Ear pinna	Tail	Faecal pellet
1. House rat	 Slender and medium-sized body	 Pointed	 Large, prominent and stands out from the body	 Longer than HB	 Black rats produce narrow cylindrical pellets with one or both ends that are pointed. Pellet colour varies with diet but is commonly a dark brown or black colour  Qty: 40 to 50 pellets daily. Size: ½” long. Shape: larger, curved, sausage-shaped with pointed ends. Found scattered

(continued)

Table 11.3 (continued)

Commensal rodents					
Species name	Body	Muzzle	Ear pinna	Tail	Faecal pellet
2. House mouse, *Mus musculus*	 Small, slender body	 Small, pointed	 Prominent, large for the size of the animal	 Longer than HB	 Spindle shaped and about ¼” size
Indian greater bandicoot rat, *Bandicota indica*	 Heavy and thick body	 Slightly pointed	 Thick, smaller for the size of the animal	 Shorter than HB	Oval and bulky
Brown rat	 Heavy and thick body	 Blunt nozzle	 Small, half buried in the fur	Shorter than HB	 Blunt and rectangular Qty: 40 to 50 pellets daily Size: ¾ inch long. Shape: larger, rectangular with blunt ends. Found in small groups

into bait stations. Generally, brown rats are exceptionally wary of unfamiliar food. Wild brown rats may even avoid a pile of wheat in an unexpected place and continue to treat it with great caution for more than a month. Moreover, when an individual rat has overcome its suspicion sufficiently to try the new food, it will only eat a small amount, perhaps 10% of its normal requirement. If it feels ill within the next 16 h or so, it will associate the illness with the ingestion of the novel food and refuse to eat it again. This phenomenon is known as aversive conditioning or 'bait shyness', normally encountered when using acute poisons (zinc phosphide). This is one of the major reasons for the success of anticoagulants as there is a delay of several days between ingestion of the bait and the onset of symptoms, thus preventing the bait toxicosis association and the development of bait shyness.

11.4.2 Feeding Behaviour

Rodent feeding behaviour varies with the species. Some rodent species eat the food from one point, whereas the others may have many feeding locations. For instance, mice may be taking food from different locations in a single foraging trip and the feed quite randomly. They exploit heavily a few feeding stations, but the sites vary from time to time. Due to the erratic feeding behaviour, the mice are considered to be one of the difficult rodents to trap or kill.

Norway rats sample the food before feeding is stabilised, and it cannot be a good indicator of food preference. Feeding stabilisation usually takes 1 or 2 days for these rodents. This sampling behaviour is seen both in young and adult brown rats; more sampling by young rats causes more spillage, but this habit leads them to food sources. The juveniles that are still suckling used to lick the paws and mouth of the mother to get the taste of the food the mother has taken. The feeding behaviour also varies among the rodents. For instance, the Norway rats hold their food in their front paws and eat them in a sitting position (Meehan 1984), whereas the mice dehusk the grain and eat.

Studies have shown the Norway rats use the trails made by other rats to find their food. Juveniles get the food cues from the mother during the nursing period which determines the dietary preference of the young rats at weaning. The juveniles observe keenly and imitate the adults while feeding and familiarise the food sources.

Mice and Norway rats prefer to feed under cover. Uncovered bait is being dragged to cover by Norway rats. This behaviour determines the success of bait stations, especially with the brown rat. Some rodent species store (hoard) food materials in their nest, and it influences the poison baiting programme. Brown rats start to hoard food after initial bouts of feeding. The hoarding depends on the type of food, number of feeding stations, distance between the source and the burrow, size of the group and sex; lactating females hoard more food than the males. Black or house rats hoard food as they do not like to feed far from their shelter.

Table 11.4 Home range of pest rodents

Sl. No.	Rodents	Home range m^2
1	*Funambulus pennantii*	65–61
2	*Rattus meltada*	88
3	*Rattus norvegicus*	459
4	*Bandicota bengalensis*	945
5	*Mus booduga*	1275
6.	Gerbils	1875

11.4.3 Home Range, Movements and Territoriality

Home Range and Movements

Home range is the space in which the rodent lives and moves on a periodic basis (Table 11.4). It is related to the territory of the rodent where it defends the area actively. Food, shelter, social effects, predation and infection determine the home ranges of rodents. Radio telemetry is used to understand the home ranges of rodents. Home range in rodent is important as it provides information on habitat utilisation and movements of rodents in a riparian habitat, which is important in the management of the species concerned. In case of mice, they tend not to move great distances. Ranges are sometimes exclusive, but often it overlaps both between and within the sexes as they also share the nest. The size of the home range of male range increases during the breeding season. Dispersal of young ones, search for food, seasonal cropping patterns and agricultural cycles determine the home range of a particular species of rodents. Home range decides the rodent movements or migration from a particular locality and their population buildup to a serious level warranting immediate control measures.

Territoriality

A territory is an area in space that is defended by an animal (by both male and female in the wild) and which contains resources (e.g. food, nest site or mates) for their survival. Territories are defended ritualistically, which minimises the need for life-threatening escalated aggression. Mice have a flexible form of territoriality. Dominant male rats aggressively defend their territory and guard a harem of females from mating by other males. Brown and roof rats used to travel 50 to 300 feet from their nests to look for resources and patrol their territory. If necessary, they can travel much farther from their territories. Normally, the patrolling is done by the dominant males during dawn and dusk time. However, the rats which are socially low-ranked individuals, who have been denied access to food by dominant rats during the night, are seen during daytime.

11.4.4 Rodent Responses to Bait Stations

Bait station is an important component in the management of pest rodents as it encourages the targeted species to sit comfortably inside and feed. It encourages the rodent to eat sufficient quantity of bait without transporting it to their burrows. Any material (wood, plastic or bamboo) and shape (cylinder is more preferred by the rats) can be used to prepare bait stations. The size of the bait station also matters; mice prefer small bait stations than large boxes or open trays for comfortable feeding. Among the various bait stations under no-choice and multi-choice conditions, house rat, *Rattus rattus*, preferred the black metallic bait stations followed by mud bait stations and showed no neophobia against these bait stations. Less bait consumption is observed when the food is offered openly (Sakthivel et al. 2013). Spacing between bait stations is also crucial for successful baiting; a spacing of 100 m is considered suitable for house rats.

11.4.5 Behavioural Resistance

Rodents tend avoid troubles, and some of behavioural characteristics of rodents make it difficult to control a rodent population especially those that had previous experience with poisoning. It is the learned behavioural characteristics which reduce a rat's tendency to eat palatable bait. It also includes the development of bait shyness and poison avoidance and enhanced neophobia. The neophobic response is more pronounced in rats that have survived. Enhanced behavioural resistance can be observed if alternative foods are available so that the rodents are not pressured to eat the poison baits. Poison avoidance behaviour, apparently heritable (physiological resistance), can also reduce rodenticide efficiency.

11.4.6 Rodenticide Resistance

Rodenticide resistance is the ability of rodents within a population to continue feeding on the rodenticide bait over a period of time, without being killed. Rats can be physiologically resistant to rodenticides, and this inheritable trait has been selected for over generations of exposure to the anticoagulant rodenticides, in commensal rodent species (Garg et al. 2017).

11.4.7 Response to Odour

Olfaction in rodent communities plays a definite role in their feeding, social behaviour and reproduction. It decides the response of rodents to traps, baits and bait stations. The odour of preferred foods and urine of opposite sex, especially the male, may increase the investigatory behaviour of rodents. Rodents depend on urine marks as a cue to detect novel objects in their territories. The traces of odour left in traps affect the behaviour of other rodents. Rodent odour varies with sex, age and reproductive stage dominant male or female or subordinate individual. Trap catches can be increased with smearing rodent scent in traps as more mice are trapped in these compared to clean traps. Some other compounds (dimethyl sulphide and dimethyl disulphide) can also be used to improve the bait acceptance and trapping of rats (Veer et al. 2002).

11.4.8 Response to Repellents

Rodents are repelled by the addition of capsaicin, citronella oil, cinnanamide, tannic acid, eucalyptus oil, etc., to baits (Spurr et al. 2001; Singla and Kaur 2014a, b; Singla and Kaur 2017; Sachdeva and Singla 2018). Denatonium is used as a deterrent for rodent gnawing. Apart from chemical substances, high-intensity ultrasonic sound is also used to repel the rodents. For want of scientific evidences on the effectiveness and the practicalities of large-scale application of these devices, limit their use in the field.

11.4.9 Grooming

Rodents keep their body neat and clean when they are at rest by grooming. They lick their body parts to remove dirt. This character of rodents can be well exploited in rodent control operations. Poisonous dust like alpha-naphthyl thiourea and anticoagulant rodenticides can be laid on rodent runways or in burrow entrance collects on the animals and is ingested during grooming.

11.4.10 Responses to Traps

Trapping efficiency varies with the species involved and the type of traps used. All rodent species are not equally trappable. Among the pestiferous rats, brown rats are comparatively difficult to trap. Intrinsic factors and previous experiences of the individual rodent also count in the trapping efficiency. Sex (male or female) and age (adult, subadult or juvenile) also determines the levels of trappability. Extrinsic factors, viz. vegetation cover, temperature and precipitation, play an important role in the relative trappability of pest rodents.

11.5 Indicators of Rodent Infestation

Rodents colonise virtually any environment that supports life. Being largely nocturnal, it is very difficult to spot the actual organism, but they leave the signs of infestation. There exist a number of tell-tale signs that rodents leave in a particular habitat (ManiChellappan and Ranjith 2019).

11.5.1 Direct Indicators

Actual spotting of live or dead rodent pest is a direct indication of rodent infestation. Unless, or otherwise, there is a population explosion, live rodent pest cannot be seen during daytime.

11.5.2 Indirect Indicators

(a) Rodent track
(b) Gnaw marks
(c) Nests
(d) Holes and marks on food packages
(e) Droppings
(f) Smudges
(g) Noise

Rodent Track
Rats, especially, establish foraging paths and rarely stray from them. As a result, tracks are often the clearly visible signs to look for.

Gnaw Marks
One of the ways to identify the rodent infestation in an area is the signs of chewing or gnaw marks close to foraging paths. As black rats are agile climbers, gnaw marks can be seen in lofts, wires, cables or other items. Shredded paper or fabric pieces on high raised places or among the clutter indicate the presence of a mouse.

Rat Droppings and Urine Stains
Droppings and urine stains are other indications of rat activity. Rodent droppings and urine are often deposited along favoured travel pathways. Rodents usually deposit their excreta in particular locations where they are frequenting. They deposit as many as 40 faecal pellets per night. The shape and size of the droppings give a clue on the species involved; black rat droppings are black tapering on both sides like a spindle or a grain of rice.

Rub or Grease Marks
Rats travel on established runways which are against any vertical surface like a wall or any solid material. They often rub their fur and smear with their body secretions against these objects. Over a period of time, these smudge marks become an indicator of rodent activity

Rat Footprints or Tracks or Runway
Rats produce a four-toed front and five-toed back footprints. They are most easily seen in muddy or dusty locations.

Rat Nests

Rats build nest and construct burrows in their habitat, viz. agricultural field bunds, in cavity walls, lofts, attics, etc. Invariably the nest consists of shredded material like grass, bark, insulation, foam, fabric and many others. Shredded paper in the house indicates the presence of rats indoor.

Rat Holes

Bandicoot and brown rats are extensive burrowers that dig out soil, concrete and wooden structures for food and nesting. The size of the burrow hole, nature of the burrow entrance and type of nesting material used are species specific.

Rodent Noise

Rodents produce a variety of sounds, which are useful to communicate with other individuals of the same species; some are audible to human ears, and some are in the ultrasonic range. Rodents make squeaks, grunts (brown rats produce grinding noise with their teeth), alarm and distress calls, and these noises have numerous behavioural responses in the receiving individuals.

11.5.3 How to Spot a Rat Infestation Indoors?

Rodents invade any space which they feel comfortable and supply the basic requirements. Since rodents are active throughout the year, periodically checking for signs of their presence is mandatory. Once rats have invaded, it is only a matter of time before the population explodes. Thorough and regular inspection for rodent infestation is obligatory to reduce the population buildup in a particular area. The following are further signs of the presence of rat inside the building:

- Rat or mouse droppings in the waste bins
- Smudge or grease marks against beams, rafters, pipes and walls
- Rat droppings around pet food storage containers
- Squeaky noises just after dusk
- Burrows beneath the compost pile or garbage can
- Rats traveling along fences at dusk
- Damaged indoor or backyard plants
- Rat carcass
- Drowned rat from swimming pool or bath tub
- Rat nests behind boxes or in drawers in the garage
- Remnants of rat nests
- Feeding marks on fruits/nuts kept exposed

11.6 Rodent Population Density Assessment

In the pursuit of rodent management, it is very important to know the population and damage caused by a particular species in question. Conducting population and damage assessments may be considered: (i) to establish the economic status of pest rodents, (ii) to determine the geographical distribution of pests, (iii) to determine the effectiveness of control measures followed and (iv) to provide information for planning.

Methods of Population Estimations

Population estimation can be done in many ways; rodent census and trapping are the most widely used and time tested ones.

11.6.1 Census by Trapping

It is also called the Lincoln index method or capture, mark and release method (CMR) where the target rodents are captured by using live traps. It is a direct method of estimating the rodent population in the field. Live traps are set in a grid manner 15 m apart in 1 ha (54 traps/ha) to capture rodents; the trapped rodents are marked and then released back in the field. Recapturing will be done after a week to estimate population using the formula:

$$\mathrm{N} = \frac{\mathrm{Mn}}{\mathrm{m}}$$

where

N = Rodent population in the study area
M = No. of rodents in first trapping
n = No. of rodents in second trapping
m = No. of marked rodents trapped

More marked individuals in recapture indicate less population in the study area and vice versa.

11.6.2 Trapline or Index Method

Index of rodent abundance can be determined by this method. Population sampling can be done by trapping on a line or lines across the study area. Snap trap or back break trap can be used for the study. Five traps should be laid in a group, and each one represents one point, which is 10–15 m apart on the line. The trapline (parallel or diagonal) should represent the entire study area. Based on the catches, trap per cent index (I) is calculated to compare the population in different areas or seasons:

$$I = \frac{M}{Xt} \times 100$$

where

X = No. of traps used in trapline
t = No. of trap set nights
M = Cumulative number of rodents trapped

11.6.3 Trapping Method

Live traps (wire box) are used to estimate the population density of rodents. The traps are loaded with very attractive bait and lay at dusk and collected back in the next morning. The trapped rodents are classified and recorded. The efficiency of trapping (trapping index) and the relative abundance of each species are calculated as population density in the study area:

$$\text{Population density} = \frac{\text{No.of rodent species}}{\text{Total rodents captured}} \times 100$$

$$\text{Trap index (TI)} = \frac{\text{No.of rodent species}}{\text{Total rodents captured}}$$

11.6.4 Active Burrow/Live Burrow Count (LBC) Method

It is an indirect method to estimate the population of rodents in the study area. The presence of rodent dropping, footprints of rodents and food material at the entrance indicate the presence of live animal in the burrow. It involves closing of all the open burrows and opening of closed burrows on the first day. The 'live burrow count' on the second day gives the approximate rodent numbers. This can be done for four consecutive days every month. The per cent live burrow count (LBC) can be worked out as follows:

$$\text{Live Burrow Count}\,(\%) = \frac{\text{No.of active burrows}}{\text{Total number of burrows counted all over}} \times 100$$

11.6.5 Food Consumption Method

Population fluctuation of rodents can be estimated by the quantity of food consumed and calculated as follows:

$$\text{Total number of rodents} = \frac{\text{Daily food conumption from a bait station}}{\frac{1}{10}\ \text{weight of the dominant rodent in the study area}}$$

(Assuming rodent’s food consumption is 1/10 of its body weight)

$$\text{Population density} = \frac{\text{Total food consumed in a month (g)}}{\text{Total food consumed over the year (g)}} \times 100$$

11.6.6 Footprint Method

Footprints on soft soil or dust can be used to calculate the rodent population in the study area. Count the number of footprints on a square unit and calculate the population as follows:

$$\text{Population density} = \frac{\text{Total no. of foot prints on randomly selected squares}}{\text{Total number of foot prints on the entire study square}} \times 100$$

11.6.7 Tracking Boards

A square or rectangular board or tile of approximately 25 × 25 or 25 × 12.5 cm on which 2–5 mm layer of dust is covered and smoothed which can be used as tracking boards to monitor rodent activities. Boards are laid out for rodent footprints near the rodent tracks or the area where the rodents are frequenting. Rodent activities are assessed based on the intensity of footprints on the tracking board (Rao et al. 2014).

11.6.8 Rodent Faeces Method

Rodent dropping is one of the vital signs of the rodent infestation and activity. Based on the physical condition of the faecal pellet, it is easy to ascertain whether the burrow is active or inactive; soft and shiny faecal pellet indicates the presence of the animal. Various sizes of rodent dropping of the same shape indicate the presence of different age groups of the rodent in that area. To estimate the rodent population, collection and counting of rodent dropping in a specified area (3m^2) is done at monthly interval. Using the following formula, the population of a particular rodent species can be calculated:

$$\text{Rodent population} = \frac{\text{Total no. of fecal pellets}}{\text{Total no.of fecal pellet of dominant rodent per day}} \times 100$$

$$\text{Population density} = \frac{\text{Total no. of fecal pellet in unit area in a month}}{\text{Total no.of fecal pelletn in a year}} \times 100$$

11.6.9 Census Baiting Method

Food consumption by rodents is used an index of their numbers in a particular habitat. Known quantity of food is offered in the infested area for about 24 h. After the exposure time, the remaining food is weighed. Again the bait stations are replenished with food and left for the next 24 h. The average daily food intake for 72 h is used to estimate the number of the rodents present in that area.

11.6.10 Other Alternate Census Techniques

(i) Rodent carcass rodenticide-treated area
(ii) Reduction in the rate of damage to the crop compared to the control plots

11.7 Rodent Damage Estimations

Rodents are persistent vertebrate pests that cause damage to agricultural crops, stored commodities and structures both by feeding and spoilage. Rodents cause immense damage to forest and plantation crops (Singleton et al. 2010; Buckle 2015). Analysis shows rodent damage is about 5–15% in rice and wheat crops alone.

The nature of rodent damage includes:

1. Direct feeding
2. Gnawing on bark of woody crops
3. Consuming seed on seed bed
4. Storing food resources in the burrow
5. Tiller cutting in cereal crops
6. Food spoilage by rodent waste
7. Soil digging and excavation
8. Damage to poultry
9. Spreading zoonotic diseases of public health and veterinary importance

11.7.1 Assessment of Rodent Damage in Rice

Rodents cause damage to rice crop at every stage of its growth:

1. Seed
2. Seedling
3. Tillering

4. Panicle and matured grain
5. Harvest
6. Postharvest and storage

Rice agroecosystems are particularly conducive to rodent infestation, and rodents are categorised as the key preharvest pest of rice in most of rice-growing areas. Grassland rats are well adapted to rice, and damage can occur right from sowing to harvest and beyond (Plate 11.6). Rat depredation in crops especially in rice could be a serious threat to food security of sizeable population of the world (Meerburg et al. 2009; Parshad 1999; Prakash 2018; Parshad et al. 2007; Rao 2003a, b). Assessment of rodent damage(s) in rice is as follows:

Preharvest Damage Assessment

Random fields are selected (five fields; six plots of 1 m^2 – four fixed plots and two variable plots), and using multistage random sampling technique, observations are taken at every stage of growth. Observations are recorded at fortnightly interval.

Rat damage in vegetative stage of rice crop – circular patches

Rat damage in vegetative stage of the crop – circular patches

Characteristic rat damage on rice tiller

Rat droppings contaminated stored rice

Plate 11.6 Rat damage in rice

Observation date/stage of crop	Affected plots (a)						Undamaged tillers (b)						Damaged tillers (c)					
	1	2	3	4	5	6	1	2	3	4	5	6	1	2	3	4	5	6

(i) **Philippine Method**
- Select a field at random.
- Select a plot at random (5–6 plots in a field).
- Select 10 rows of plants for observation.
- In each row, select randomly 10 hills at equal distance.
- In each hill, count rat cut tillers and other healthy tillers.
- Tabulate for 100 (10 rows x 10 hills) observations.
- Calculate damage incidence, $P = \frac{A \times C}{B + C.}$

  where

  A = Number of damaged hills
  B = Number of undamaged tillers in A
  C = Number of damaged tillers

(ii) **Diagonal Method**

Select a field at random.
- Select a plot at random.
- Select a diagonal.
- Start with first hill.
- Count cut and uncut tillers.
- Select next position on the diagonal.
- Repeat steps 5 and 6 until 25 samples at equidistance are taken.
- Calculate damage incidence (P):

$$P = \frac{A \times 100}{(A + B)}$$

where

A = No. of damaged tillers
B = No. of healthy tillers

Per cent vegetative loss can be calculated as

$$\%\text{loss} = \frac{a \times c}{b + c}$$

where

a = Number of affected plots
b = Number of healthy tillers
c = Number of damaged tillers

Damage at the time of tillering also contributes to the actual loss at the time of harvest as those tillers regenerate and produce poor-quality grains. Yield from such tillers is deducted from the total yield, and the yield loss can be worked out as follows (Raj 2018).

Total no. of plants N	No. of damaged plants $N1$	No. of healthy plants $N-N1$	Yield of grains from health plants (g) Y	Average yield in g $\frac{Y}{N-N1} \times N$	Loss in yield (g) $\frac{(y)}{(N-N1\times N)-Y}$

Hoarding Loss Assessment

Rice field rodents store a large quantity of grains in their nest and cause severe preharvest losses. The loss assessment can be done as follows.

Observation date/stage of crop	No. of rat burrow	No. of burrow/ha	Food chamber			Nature of food store
			Without food (No.)	With food (No.)	Quantity	

$$\text{Food hoarding/ha} = \frac{\text{Average Amount}}{\text{Food stored}} \text{Average of number of burrow systems per ha}$$

(i) **Mean number of burrow systems per ha**
  (i) Select field at random.
  (ii) Count the number of burrow/ha.
  (iii) Count the number of burrow openings/systems.
  (iv) Dig out the burrow system and note the number of food chambers with or without hoarded food.
  (v) Measure the amount hoarded at least from 5–6 fields.
  (vi) Record data and calculate the average and hoarding losses/ha as follows:

Date/stage of crop	No. of burrow openings	No. of burrow systems/ha	No. of food chambers	Food eaten/intact	
			Without food	With food	Amount of food stored (g)

11.7.2 Assessment of Rodent Damage in Sugarcane

Sugarcane ecosystem, which is stable and undisturbed for nearly 3 years, provides ideal place for food and shelter for small rodents. Rats cause direct loss of cane by eating into the internodes of standing and lodged cane. This permits entry of insects and pathogens and also causes physiological stress. Damage may also come from the eating of growing tissues and of underground parts by fossorial rodent species. Loss relates to the proportion of damaged canes, and it is around 10–20% of its sugar content. The proportion of damaged canes is often high, up to 90% or more, with a significant proportion dying.

In the Indian subcontinent, sugarcane is an important crop subject to heavy rodent damage. The most common rodent pest is *B. bengalensis*, which has caused damage as high as 63% of canes in Andhra Pradesh (Rao 2003a, b). Other species damaging sugarcane in India are squirrels and porcupines (Srivastava 1992; Wood and Singleton 2015).

Damage caused by rodents to sugarcane is assessed similar to rice crop.

Date and crop stage	No. of affected plots (a)						No. of undamaged tillers (b)						No. of damaged tillers (c)					
	1	2	3	4	5	6	1	2	3	4	5	6	1	2	3	4	5	6

$$\text{Loss} = \frac{(a \times c)}{(b + c)}$$

where

a = Number of damaged plots
b = Number of unaffected tillers
c = Number of affected tillers

11.7.3 Rodent Damage Assessment in Coconut

Coconut palms of all ages are being attacked by rodents, and the developing nuts damaged fall, prematurely (Plate 11.7). Since natural ‘thinning out’ takes place in coconut palms, assessment of actual losses is not straightforward. In India, Bhat (1992) showed losses varying between geographical locations (8.7% in Andhra Pradesh to 50% in the Lakshadweep Islands). The main species include various sub-species of *Rattus*, in particular *R. r. wroughtoni, R. r. rufescens* and Malabar giant squirrel (*Ratufa indica*) in mainland India (Rao et al. 1984; ManiChellappan and Vidya 2018) and *R. r. andamanensis* and *R. r. holechu* in the Andamans.

Nursery and young field palms may also be attacked by the ground-dwelling rodents, *B. bengalensis*, *B. indica* and *Tatera indica*. The porcupine, *Hystrix indica,* also attacks at this stage (Wood and Singleton 2015).

In a plantation, 10 palms out of 100 palms may be randomly sampled for nut damage once in a month. Care may be taken to count the freshly damaged ones. The damage can be worked out as follows:

Plate 11.7 Rodent damage in coconut

Rat damaged coconuts

Malabar giant squirrel damaged coconuts

Date	Number and stage of nut damaged	Remarks
	Tender/medium/mature	Rodent species

Select a block of plantation with 100–150 palms; identify and count the number of infested palms on the basis of freshly fallen damaged nuts near the palm base. Calculate per cent rodent-infested palms (RI) using the formula:

$$\mathrm{RI}\ (\%) = \frac{\text{Number of palms with fallen nuts}}{(\text{Total number of palms})} \times 100$$

For assessing per cent nut damage, select 10 palms with fallen nuts in the same orchard and record the number of healthy and damaged nuts on the selected trees covering all branches:

$$\text{Nut damage}\ (\%) = \frac{\text{Number of damaged nuts}}{\text{Total number of nuts}} \times \mathrm{RI}$$

11.7.4 Rodent Damage Assessment in Oil Palm

Rats gnaw the unripe bunches, even through to the kernels, leaving characteristic scarring and the ripe fruitlets detached from the bunch. Rats also feed on those detached fruitlets and often carry them away. A loss of about 5% to 10% yield is reported from oil palm plantations (Wood and Singleton 2015).

Young palms may be gnawed at the base by rats before they begin to fruit. The pests occasionally penetrate the bud and kill the palm, altogether. Hystricomorphs are conspicuous pests in the young plantations. In the Far East, the Malayan porcupine, *Hystrix brachyurus*, can destroy large numbers of palms, though this is usually confined to the locality near its habitat in secondary jungle or scrub.

Fresh fruit damage in oil palm is worked out by counting undamaged and damaged fruits in each palm by the formula:

$$\text{Fresh fruit damage}\ (\%) = 100\left(\frac{\mathrm{a}}{\mathrm{b}}\right)$$

where

a = No. of palms with fresh fruit damage and
b = No. of palms assessed

11.7.5 Rodent Damage Assessment in Cocoa

Rodents bore into the cocoa pods, and large-bodied species can take whole beans, while small ones may feed only on the mucilage that surrounds the beans. The proportion of holed pods can be very high, and they become increasingly susceptible to damage as they ripen (Plate 11.8). The damage is compounded by ensuing fungal infection, and affected pods are all lost. Cocoa grown under coconuts is susceptible to attack by both rats and squirrels. Persistent widespread damage to cocoa is likely near to borders with crops that support rat, viz. oil palms or rice, where losses may reach even 100%. Heavy losses can occur in India by Western Ghats squirrel,

Plate 11.8 Rodent damage in cocoa

. Rodent damaged cocoa pods

Funambulus tristriatus, the south Indian palm squirrel (*F. palmarum*) and black rat, *Rattus wroughtoni* (Bhat 1992; Baco et al. 2010).

Since rodent damage is restricted to ripe pods, it may be categorised as healthy or undamaged, scratched and freshly bitten pods. Per cent pod damage is calculated as:

$$\text{Cocoa pod damage }(\%) = 100\left(\frac{b+c}{a+b+c}\right)$$

where

a = Number of unaffected pods
b = Number of pods with scratches
c = Number of freshly bitten pods

11.7.6 Rodent Damage Assessment in Groundnut, Soybean, Pineapple and Cardamom

Squirrel *Funambulus pennant* is reported to remove 25% of planted groundnut seeds in India, and the summer crop may be left unplanted. A strong relationship of rat activity to the suitability of the surrounding vegetation is noted. All stages of soybean in India are subject to damage, mainly by *Millardia meltada* and also by *R. rattus* and *B. bengalensis*.

Up to 16% of ripening pineapples are damaged by *Bandicota bengalensis* and *R. rattus* in Bangladesh in a range of study plots. In commercial plantings in India, cardamom seed capsules are damaged by a range of fossorial rodents, gerbils and squirrels (Srihari and Chakravarthy 1992), in particular *B. bengalensis*. Seed capsules may be emptied, over 12% in the worst cases. The plants too can be damaged, especially in the young stages.

11.7.7 Rodent Damage Assessment in Store

The problems caused by rodents in stores are wide ranging. Broadly, losses may be attributed to the following:

(a) Direct consumption of food
(b) Food contamination and damage
(c) Structural damage
(d) Disease transmission
(e) Source of reinfestation of adjoining areas
(f) Cost associated with control operations

Many estimates lie in the range 1–10% and invariably include total losses due to consumption as well as spillage, damage and contamination, resulting from rodent activity (Smith and Meyer 2015). Some estimates are that up to ten times as much food is lost as a result of spillage and contamination as is lost to direct rodent consumption.

11.8 Rodent-Borne Diseases

Rodents are carriers of many zoonotic diseases to human and animals. These diseases can be categorised into two (Fig. 11.4): (i) transmission of pathogen through rat bite and excreta and (ii) transmission through intermediate arthropod vector (rat flea, ticks or mites) (Meerburg et al. 2009) (Tables 11.5. and 11.6). Over the last millennium, it has been estimated that rat-borne diseases may have taken more lives than all of the wars ever fought (Battersby 2015).

Rat-bite fever is caused by the casual and active rat bites and scratches, while the bacteria in rat urine causes leptospirosis (Fig. 11.5). Lymphocytic choriomeningitis

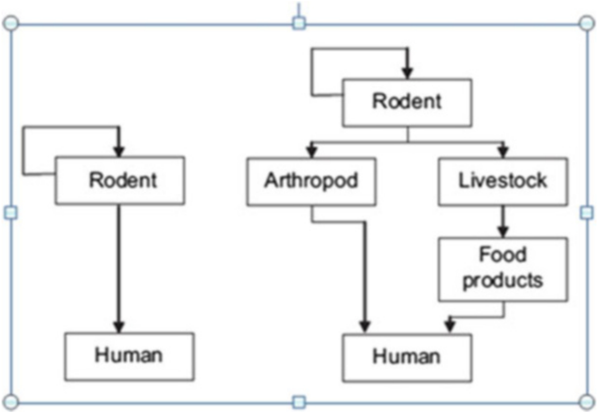


Fig. 11.4 Transmission of rodent borne disease to human

Table 11.5. Major rodent-borne zoonotic diseases

S. No.	Causative organism	Number of diseases
1.	Bacteria	19
2.	Virus	22
3.	Sprozoea/protozoa	3
4.	Zoomastigophorea	3
5.	Cestoda/flatworm	3
6.	Nematoda	6
7.	Lobosea/amoeboid eukaryotes	1
8.	Conoidasida/alveolates	1

(LCMV), a viral infectious disease, is transmitted through the saliva and urine of rats. Rat flea bite transmits plague, one of the most historically dangerous diseases. The Black Death is widely thought to have been an outbreak of bubonic plague that transmuted into the pneumonic form.

Rats also are a potential source of allergens. Rat droppings, flakes of skin (dander) and rat hairs can cause allergic reactions.

11.9 Management of Pest Rodents

Being herbivore, the choice of food for rodents is unlimited. No crop is spared by these small vertebrates at any stage of its growth. The extent of crop damage depends on the rodent species involved, location and stage of crop, rodent population level and availability of shelter. Devastations have been well documented in food crops, pulses, oilseeds, plantation crops, fruit orchards, grasslands and forestry plantation crops. Surge in rodent population leading to outbreak may be due to prolonged dry

Table 11.6 Overview of different pathogens that may be transmitted by rodents

Disease	Agent	Carrier/ reservoir	Population at risk	Chance	Human health	Economy
Hantavirus pulmonary syndrome	Virus, Bunyaviridae	Carrier	2	1	3	1
Haemorrhagic fever with renal syndrome (+ other haemorrhagic fevers)	Virus, Bunyaviridae	Carrier	2	2	2	2
Nephropathia epidemica	Virus, Bunyaviridae	Carrier	1	1	1	1
Crimean-Congo haemorrhagic fever	Virus, Bunyaviridae	Reservoir	1	1	3	1
Borna disease	Virus, Bornaviridae	Reservoir	1	1	1	2
Omsk haemorrhagic fever	Virus, Flaviviridae	Reservoir	1	1	1	1
Kyasanur forest disease	Virus, Flaviviridae	Reservoir	1	1	1	1
Apoi virus disease	Virus, Flaviviridae	Unknown	Unknown	Unknown	Unknown	Unknown
Tick-borne encephalitis	Virus, Flaviviridae	Reservoir	2	1	3	1
Powassan encephalitis	Virus, Flaviviridae	Reservoir	1	1	1	1
Lymphocytic Choriomeningitis virus (LCMV)	Virus, Arenaviridae	Reservoir	1	1	1	1
Lassa fever	Virus, Arenaviridae	Carrier	2	2	3	2
South American arenaviruses (Junin, Machupo, etc.)	Virus, Arenaviridae	Carrier	2	2	3	1
North American arenaviruses	Virus, Arenaviridae	Carrier	1	1	Unknown	Unknown
Colorado tick fever	Virus, Reoviridae	Reservoir	1	1	1	1
Venezuelan equine encephalitis	Virus, Togaviridae	Reservoir	2	2	2	2
Western equine encephalitis	Virus, Togaviridae	Reservoir	1	1	1	1
Hepatitis E	Virus, Caliciviridae	Reservoir	1	1	1	1
Cowpox	Virus, Poxviridae	Reservoir/ carrier	1	1	1	1
Contagious viral animal diseases (classical swine fever, foot and mouth disease)	Virus, Picornaviridae (FMD); Flaviviridae (CSF)	Reservoir?	0	1	0	3
Leptospirosis (Weil's disease)	Bacteria, Spirochaetes	Carrier	2	2	3	2

Lyme disease	Bacteria, Spirochaetes	Reservoir	3	2	1	2
Tick-borne relapsing fever	Bacteria, Spirochaetes	Reservoir	2	1	1	1
Scrub typhus	Bacteria, Alphaproteobacteria	Reservoir	2	1	3	1
Murine typhus	Bacteria, Alphaproteobacteria	Reservoir	3	1	1	1
Sylvatic epidemic typhus	Bacteria, Alphaproteobacteria	Reservoir	1	1	1	1
Queensland tick typhus or spotted fever	Bacteria, Alphaproteobacteria	Reservoir	1	1	1	1
Rocky Mountain spotted fever	Bacteria, Alphaproteobacteria	Reservoir	1	1	3	1
Rickettsialpox	Bacteria, Alphaproteobacteria	Reservoir	2	1	0	1
Bartonella illnesses	Bacteria, Alphaproteobacteria	Reservoir	2	2	1	1
Disease	Agent	Carrier/ reservoir	Population at risk	Chance	Human health	Economy
Human granulocytic anaplasmosis	Bacteria, Alphaproteobacteria	Reservoir	2	1	1	1
Q-fever	Bacteria, Gammaproteobacteria	Reservoir	3	2	3	2
Salmonellosis	Bacteria, Gammaproteobacteria	Carrier	3	1	1	3
Tularemia	Bacteria, Gammaproteobacteria	Carrier	2	1	3	1
E. coli 0157/VTEC	Bacteria, Gammaproteobacteria	Carrier	2	1	3	2
Plague (Yersinia pestis)	Bacteria, Gammaproteobacteria	Reservoir	2	2	2	2
Campylobacteriosis	Bacteria, Epsilonproteobacteria	Carrier	3	1	1	3
Rat-bite fever and Haverhill fever	Bacteria, Fusobacteria	Reservoir	2	1	3	1
Listeriosis	Bacteria, Bacilli	Carrier	3	1	3	2
Toxoplasmosis	Parasite, Sporozoea	Reservoir	3	2	2	3
Babesiosis	Parasite, Sporozoea	Reservoir	3	2	1	1

(continued)

Table 11.6 (continued)

Disease	Agent	Carrier/ reservoir	Population at risk	Chance	Human health	Economy
Cryptosporidiosis	Parasite, Sporozoea	Reservoir	3	2	1	3
Chagas disease	Parasite, Zoomastigophorea	Reservoir	3	1	3	2
Leishmaniasis	Parasite, Zoomastigophorea	Reservoir	3	2	3	2
Giardiasis	Parasite, Zoomastigophorea	Reservoir	3	2	1	2
Taeniasis	Parasite, Cestoda	Reservoir	1	1	1	1
Rodentolepiasis	Parasite, Cestoda	Reservoir	1	1	1	1
Echinococcosis	Parasite, Cestoda	Reservoir	2	1	3	1
Schistosomiasis	Parasite, Trematoda	Reservoir	3	2	1	3
Human fascioliasis	Parasite, Trematoda	Reservoir	3	1	1	3
Brachylaimiasis	Parasite, Trematoda	Reservoir	1	1	2	1
Alariasis	Parasite, Trematoda	Reservoir	1	1	0	1
Echinostomiasis	Parasite, Trematoda	Reservoir	1	1	0	1
Trichinosis	Parasite, Nematoda	Reservoir	3	2	1	2
Capillariasis	Parasite, Nematoda	Carrier	3	1	1	1
Angiostrongylosis	Parasite, Nematoda	Reservoir	2	1	3	1
Toxocariasis	Parasite, Nematoda	Carrier	1	2	0	2
Baylisascariasis	Parasite, Nematoda	Carrier	1	2	1	2
Aelurostrongylosis	Parasite, Nematoda	Reservoir	0	0	0	0
Amoebic dysentery	Parasite, Lobosea	Reservoir	3	1	3	1
Neosporosis	Parasite, Conoidasida	Reservoir	0	1	0	2

Reservoir: rodents harbour disease-causing organisms and thus serve as potential sources of disease outbreaks, but always via a vector (tick, sand fly, etc.)
Carrier: rodent that shows no or limited symptoms of a disease but harbours the disease-causing agent and is capable of passing it directly onto humans
Population at risk: focal = 1, regional = 2, more than 2 continents = 3
Chance: chance of contracting the disease (all pathways, not only via rodents): small chance = 1, moderate chance = 2, high chance = 3 Human health: mortality without treatment <5% = 1, 5 to 10% = 2, >10% = 3. No mortality = 0
Economy: losses in terms of morbidity combined with other losses (e.g. in animal productivity): small losses = 1, moderate losses = 2, huge losses = 3

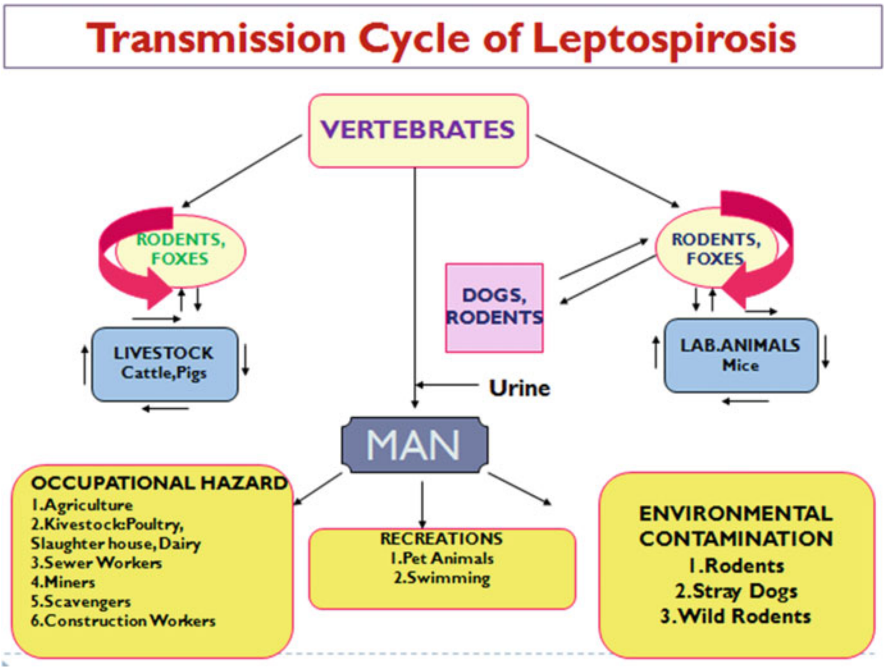


Fig. 11.5 Transmission cycle of leptospirosis

spell followed by heavy rains, failure of monsoon in preceding years, flash flood and bamboo flowering (in certain places).

A very well-planned strategy is required for an effective and cost-effective rodent management programme on any scale; meticulous planning is required to have long-term effective population reduction. Survey, different control methods (ecological, physical, mechanical, biological and chemical), record keeping, monitoring and review are some of the essential elements of an integrated rodent pest management (IRPM) programme (Rao 2019).

11.9.1 Survey

Prior to the full-scale practical management programme, it is essential to know the extent of infestation to be treated, identification of the targeted species, the activity pattern of the targeted species, sheltering materials, available food sources, nontarget risks and target sites are known from the initial survey. The survey also aims to identify the rodent species through indirect evidences of rodent activities. New technologies of survey include the options based on electronics and information technologies (real-time data recording with motion detectors) enabling constant remote monitoring.

11.9.2 Environmental Management

The presence of rodent infestation indicates suitability of that habitat which provides everything to the rodent to survive, viz. food, water they need and shelter to live, move around and breed successfully. Unavailability of these factors lowers the carrying capacity of that habitat that in turn reduces the level of rodent activity. For an effective programme, these factors are critical, and modifications of some of these key factors ensure no reinfestation in the same area in the near future once the control measures are withdrawn.

(i) **Cultural practice:** An ecological technique to create stress among the native rodent pests by manipulating the habitat. It involves low cost and just modification in crop husbandry practices, like deep ploughing, removal of wild vegetation and refuse of previous crops.
(ii) **Harbourage removal**: rodents are wary of open places, and they require some sort of cover for their movement, foraging, etc. Removing the potential harbourage makes the target rodent species vulnerable.
(iii) **Rodent-proofing at home**
    - Mending damages in ventilator screen, around foundation and on walls in time
    - Tight-fitting cover for the crawl spaces
    - Repairing all gaps and crevices around the inlets
    - Rat-proofing of windows with screens
    - Tight-fitting doors

(i) **Use of Repellents**

Capsaicin and denatonium have repellent property and prevent rats from gnawing objects. Other compounds added to bait to reduce palatability are cinnanamide and tannic acid (Spurr et al. 2001; Singla 2013).

(ii) **Use of Antifertility Agents**

A host of chemicals with antifertility effect on pest rodents have been identified, and some of these are anti estrogen V-ll, diphenylmethane derivatives, metepa, tepa, tetradifon, furadantin, colchicine, etc.

(iii) **Use of Antifeedant and Antifertility Compounds**

This is one emerging area in rodent pest management. They include a host of chemicals with antifertility effect on pest rodents that have been identified, viz. anti-estrogen V-ll, diphenylmethane derivatives, metepa, tepa, tetradifon, furadantin, etc. (Tripathi and Chaudhary 2005), and many plant products with antifeedant and antifertility properties have been identified (Singla 2013).

(iv) **Trunk Banding**

Trunk banding with a galvanised iron or zinc or self-adhesive polyethylene sheet of the size 2 × 2 feet is wrapped around the coconut palms at a height of 6 feet that reduces damage to nut by palm-climbing rats.

(v) **Electronic Rat Repellents**

Devices which produce unfamiliar frequencies apparently scare the rodents. However, like other vertebrates, rats accustom to the electronic resellers.

(vi) **Electric Fencing**

Low volt current passed along the metal cable that protects the agricultural fields or plots from the marauding rodents. It is an efficient deterrent against not only rodent but also against other vertebrate pests. Animals that experienced the shock try to avoid the area fenced.

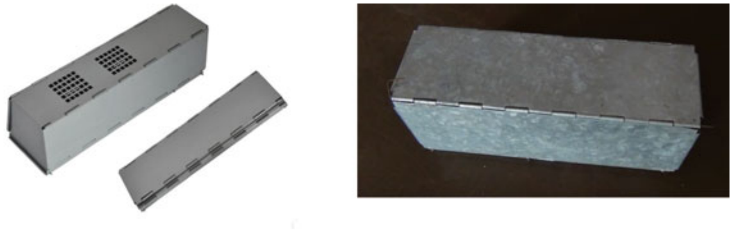
11.9.3 Rodent Population Control

Rodent pest management is always tricky and needs prior scientific knowledge about rodents. Rodent trapping is one of the direct methods of controlling rodent population and can be done in two ways: (i) kill trapping and (ii) live trapping. There are many methods available by which pest rodents are either caught or killed at once.

11.9.3.1 Physical Control

Physical control techniques include live and kill trapping, ultrasound, electromagnetic fields, shooting, etc.

Various kinds of traps (Plate 11.9a, b) are used to remove rodents from their habitat. Trapping also provides details about the species composition and population density of the rodent pest and hence is useful in monitoring purposes. Trapping is considered as the safest killing method around human habitations. Traps can be used for a prolonged period, cost-effective and safe but more labour intensive. Both intrinsic factors concerned with the target species like the species, previous experience and age of the rodents and the extrinsic factors like temperature, cover and moisture determine the efficiency of the traps (Drickamer et al. 1999; Davis et al. 2003).



Collapsible rat trap **Sherman trap**

Box trap **Live rodent trap**

Multi catch rodent trap **Moncompu trap**

Plate 11.9a Rodent live traps

11.9.3.1.1 Live Trapping

(i) Use of Live Traps

Live traps are not always preferred in the large-scale rodent management programmes as it involves killing of trapped rodents subsequently. However, for conservation purposes and monitoring of rodent population, live traps are being used. In multiple-catch live traps, as many as 20 rodents can be trapped.

(ii) Sherman Traps

Sherman traps are standard live traps used worldwide. It is a box trap with collapsible arrangement and can be safely handled. These traps are used to trap

Kanimangalm rat trap

Snap trap

Glue trap/ glue board

Bandicoot Snap trap

Bow trap

Trap laid in field

Plate 11.9b Rodent kill traps

rodents for population studies, collection, teaching, mark-recapture-release programmes, etc.

(iii) **Box Trap**

Wooden, plastic or metal box traps are widely used in household to capture the commensal rodents using attractive bait materials.

11.9.3.1.2 Use of Kill Traps

(i) Snap Trap (Break Back Trap)

Traps are made up of either wooden or tough plastic or metallic one. Efficiency of the trap can be increased by pre-baiting the rodents for 1 or 2 days; any fried material or a piece of coconut meat can be used as a pre-bait material. Care should be taken as it inflicts wounds if not properly handled and hence is not safe for kids and pets when used indoors. These traps can be used in the fields either linearly or diagonally.

(ii) Bow/Arrow Trap

These are fabricated from bamboo in the shape of arrow or bow. These are very effective against rice field rodents and used in Tamil Nadu, Andhra Pradesh, northeastern states of India. It needs some skill to use these traps.

(iii) Glue Board/Glue Trap

Sticky material is used to trap small rodents like mouse, and these traps work with the same principle of flypapers. Like the live traps, the trapped animal has to be killed which makes these traps less attractive for large-scale rodent control programmes.

(iv) Electrocution Trap

Rodents are killed by electrocution, and these are expensive traps. These traps require frequent checking for dead rodents. Though the traps are excellent in getting the problem rodents, these traps have some inherent drawbacks. Trapping is usually not practical if rodents are numerous, affected areas are too large, traps are costly or reinvasion is rapid. If traps are used, the intensity of efforts needs to be related to the numbers and activity of rodents and compared with the level of crop damage. Usually, trapping has proven to be so labour intensive that little benefit is achieved or efforts cannot be maintained because farmers lose interest, when local rodent activity is low before crops are susceptible. Still, in some special situations, for example, experimental fields of deepwater rice (Islam and Karim 1995: Huan et al. 2010), trapping has been used effectively to manage rat damage.

Trap barrier system: Trap barrier system is an eco-friendly rodent management method. It has been proved successfully in managing rice field rats (Singleton and Bell 2002; Rao and Kishore 2010)

Indigenous Methods to Kill Rodents

Numerous local methods are being used to kill rodents at home and in the agricultural fields. This indigenous technical knowledge (ITK) of rodent control includes the use of leaf and bark of *Gliricidia* mixed with rice.

Barn owl in the natural habitat

Artificial nesting site for the barn owl

Inverted coconut petiole perch for barn owl

T-perches for barn owl

Plate 11.10 Barn owl – rodent biocontrol agent

11.9.3.2 Biological Methods

Encouraging natural predators of problematic rodents is gaining importance as acute rodenticide causes secondary poisoning in nontargeted organisms. The role of barn owl in the management of rodents in rice fields and coconut plantations is immense (Neelanarayanan 1996; ManiChellappan et al. 2019; Roulin 2020). Barn owl is encouraged to breed artificial nesting sites established in the crop fields that are of immense value in managing field rats (Plate 11.10). Owls are encouraged in agricultural fields by providing T-perches (@ 25/ha) and inverted coconut petiole (@50/ha).

11.9.3.3 Chemical Method

For vertebrate pest management, naturally occurring compounds (cyanide and strychnine) have been used a number of years, globally; the same is true with zinc phosphide. Most of the research on rodenticide happened between 1940 and 1990. Sodium fluoroacetate is the salt form of fluoroacetate. Fluoroacetate is the toxic component of 1080 (developed in the 1940s), which occurs naturally in some poisonous plants. Anticoagulant rodenticides can be broadly classified in acute and subacute blood anticoagulants and fumigants (Endepols et al. 2015). Time and method of application of these rodenticides play an important role in getting desired effects on the rodent pest management (Chaudhary and Tripathi 2005, 2019).

11.9.3.3.1 Acute Rodenticides

These are the substances that cause death of rodents within a shortest time of exposure to the rodenticide either from a single dose or from multiple doses for a short period. Acute rodenticides cause death as a result of:

- Paralysis of central nervous system
- Respiration failure
- Cardiac irregularities
- Gastrointestinal irritation and haemorrhage
- Destruction of red blood cells
- Destruction of kidney and liver tissues
- Blocking of citric acid oxidation
- Hypothermia

Alphachloralose is an acute toxicant which induces hypothermia and death when used as baits (2–4% of the active material). However, the use of this compound is restricted due to its bird toxicity.

Zinc phosphide is another most widely used acute rodenticide throughout the world since 1911 (first used in Italy). Clinical signs of poisoning appear within 15 minutes after the ingestion and death within 3–12 h. Zinc phosphide reacts with the HCl in the stomach and releases phosphine which causes respiratory and heart failures which leads to death. It is effective against both urban and field rats (2%). At higher concentrations, it is less acceptable to rodents. Its garlic-like odour appears to be attractive to rodents unless bait shyness has developed, whereas this smell tends to repel higher animals.

Sodium fluoroacetate (1080) occurs naturally in the seeds of *Dichapetalum braunii* that contain levels of fluoroacetate up to 8000 mg/kg. It is very effective against many vertebrate pests. Clinical symptoms of poisoning appear within 30 minutes of its consumption, and death occurs within 24 h by inhibiting the energy production from tricarboxylic acid (Krebs) cycle.

Other acute rodenticides include norbormide, thallium sulphate, strychnine and red squill.

11.9.3.3.2 Subacute Compounds

These compounds are with delayed action. There are two compounds under this category – bromethalin and cholecalciferol.

Bromethalin: It is a single-dose rodenticide developed in the 1970s and predominantly used as bait against commensal rodents. It is used in baits (@ 0.005 or 0.01%)

Cholecalciferol (vitamin D3): It is used in baits (0.1%) for the control of rodents. It causes hypercalcaemia by mobilising calcium from bones, and death occurs within 3–7 days. It has low secondary poisoning on nontarget species like birds. Low doses of cholecalciferol are being added to anticoagulant-containing baits to increase their effectiveness.

11.9.3.3.3 Chronic Rodenticides or Blood Anticoagulant Rodenticides

Anticoagulants interfere with the blood clotting mechanism in the target species, rupture fine blood capillaries and cause internal haemorrhage, leading to death. Used in very low concentration, these rodenticides cause death after several days, and hence the development of bait shyness to anticoagulants is remote.

Anticoagulant rodenticides fall into two groups, first- and second-generation anticoagulants, and these rodenticides cause internal haemorrhage and death.

First-generation rodenticides: It includes warfarin, chlorophacinone and diphacinone that are multidose anticoagulants. Death occurs after ingesting the bait for a period of several days.

Warfarin: It is a hydroxyl coumarin compound first introduced in 1947; warfarin interferes with the blood clotting factors in the target animal and leads to death.

Diphacinone: It is an indane-dione class compound having comparatively more toxicity to rats and mice.

Chlorophacinone: It is similar to diphacinone but with more toxicity.

Coumatetralyl: It has more toxicity than warfarin and can be used in multiple ways, viz. tracking powder or bait, wax block or paste for rodent control.

Second-generation anticoagulants: It includes brodifacoum, bromadiolone, difenacoum, difethialone and flocoumafen which are single-dose anticoagulants and have greater affinity for vitamin K-epoxide reductase. However, the field use of second-generation anticoagulants has resulted in reports of adverse effects on wildlife.

Though the second-generation anticoagulants are used widely against commensal and to a certain extent field rodent management, their persistence on rodent's body than do the first-generation anticoagulants is one of the environmental concerns. They may pose potential hazards to predators and scavengers, which may consume poisoned rodents. Hence, proper disposal of dead rodents and application of these rodenticides as per the label directions may reduce the secondary poisoning in nontarget animals.

11.9.3.3.4 Fumigant Rodenticides

Fumigants are used to manage rodent in situations where the use of other rodenticides is either ineffective or impractical. Fumigant rodenticide application requires extreme care as the poisonous gases emanating from the compounds are deadly.

Aluminium phosphide: This is one of the most widely used fumigant, which releases phosphine (PH_3) gas when it reacts with moisture. It is used in storage to manage insects and rodents. It is available in either pellets or tablets and for burrow fumigation. After insertion of the fumigant pellets/tablets into the burrow, the burrows are closed with moist soil that results in the production of phosphine that becomes toxic to the rodents in the burrow.

Cyanogas: It is an alternative to acute rodenticide which is normally delivered into the burrow using a fumigator.

Carbon dioxide (CO_2): It is very effective against commensal rodents like mouse. Research shows that rats become unconscious in 25 seconds when exposed to 100% CO_2. Carbon dioxide offers many benefits as a fumigant. First, the gas is heavier than air, so it naturally sinks into the burrow. Second, it is noninflammable. Finally, it offers a greater margin of safety for the applicator than other fumigant gasses.

Ignitable gas cartridges: Ignitable gas cartridges are one of the burrow fumigants. The USDA-APHIS manufactures two gas cartridges, which contain sodium nitrate and charcoal as active ingredients that when lit produce toxic carbon monoxide gas, which kills the occupants of the burrow.

Smoke: Hand-operated centrifugal blower-type burrow fumigator using ignited straw as the source of smoke is being used in the rice-growing areas in India for burrow fumigation. The effectiveness is almost near to the aluminium phosphide fumigation (Reddy et al. 2005).

Estimates showed that burrow fumigation costs five to ten times more than toxic rodent baits; the benefit of burrow fumigants, however, is that they lack residual toxicity and have no secondary poisoning on other animals that reoccupy the treated burrows or feed on carcasses killed by the fumigants. Fumigants will be an appropriate method after the rodent baiting programme to deal with the residual rodent population still holed up in the burrows.

Bait Placement and Bait Stations

Rodenticides are highly toxic and must be handled according to the directions. These rodenticides can be used as bait, contact toxicant or fumigants. To improve the safety and effectiveness, bait stations are recommended. Keeping bait stations in places frequented by the rodents or near the burrows and harbourage improves the effectiveness of both the bait and bait stations. Both oil and sugar in the bait increases its attraction to the rodents. Leftover bait and carcasses of poisoned rodents should be properly disposed of.

11.9.3.3.5 Other Control Methods

Flooding the burrows of field rodents is an effective method. Using a water hose, water can be pumped into the burrows to kill the inhabitants.

Rodenticide Dust or Tracking Powder

Grooming behaviour is exploited to manage rodents in certain situations. Tracking powders like alpha naphthyl thiourea (ANTU), chlorophacinone, diphacinone, zinc sulphide, coumatetralyl, etc., are some of the tracking powders used in the rodent management. By placing these rodenticide powders in or near the rodent runways and burrows, rodents pick up the toxicant on their body. While grooming action, the chemicals get into the system. If properly used, rodenticide powders are safe to nontarget organisms.

11.9.3.3.6 Rodenticide Evaluation

New rodenticides are evaluated for their toxicity on rodents in laboratory.

Oral Toxicity

Determination of oral LD_{50} (mg/kg) is done by oral administration of technical grade and by stomach intubation technique.

Baseline Susceptibility

Susceptibility of rodents to rodenticide can be studied by plotting log days feeding against probit mortality. Then the Lethal Feeding Period (LFP) to kill 50% of the experimental rats can be determined.

No Choice Feeding Tests

In No Choice Feeding experiments, rodenticide at different doses mixed with bait is given to individually caged rodents to record the number of days taken from the initial feed to death of the rodents.

Choice Tests

Along with rodenticide laced food, plain food is also provided to the caged rodents to see if it is acceptable to the animals and mortality days to death are recorded.

Experiments are conducted to know the effectiveness of the rodenticide in field conditions based on live burrow count (before and after the poison baiting) and crop damage index.

11.9.3.3.7 Warning on the Use of Rodenticides

Pesticides are poisonous. Always read and carefully follow all precautions and directions provided on the container label.

Pets and Rat Control

Rodent control methods and materials are equally affecting the pets (dogs and cats) as well. Secondary poisoning of pets and wildlife is possible if they eat rodenticide-poisoned rats. It is always better to keep pets away from the leftover bait and dead or dying rodents.

11.9.3.3.8 Rat Control Campaign

It involves meticulous planning and neat execution. The campaign should be conducted during (i) preharvest stage of the crop and (ii) nonbreeding period (Fig. 11.6). Residual rodent population if not controlled would contribute to their population (Chaudhary and Tripathi 2005; Raj and Naik 2015). The operations include precontrol, control and postcontrol operations. The initial rodent populations are estimated by live burrow count and damage index. The difference of precontrol

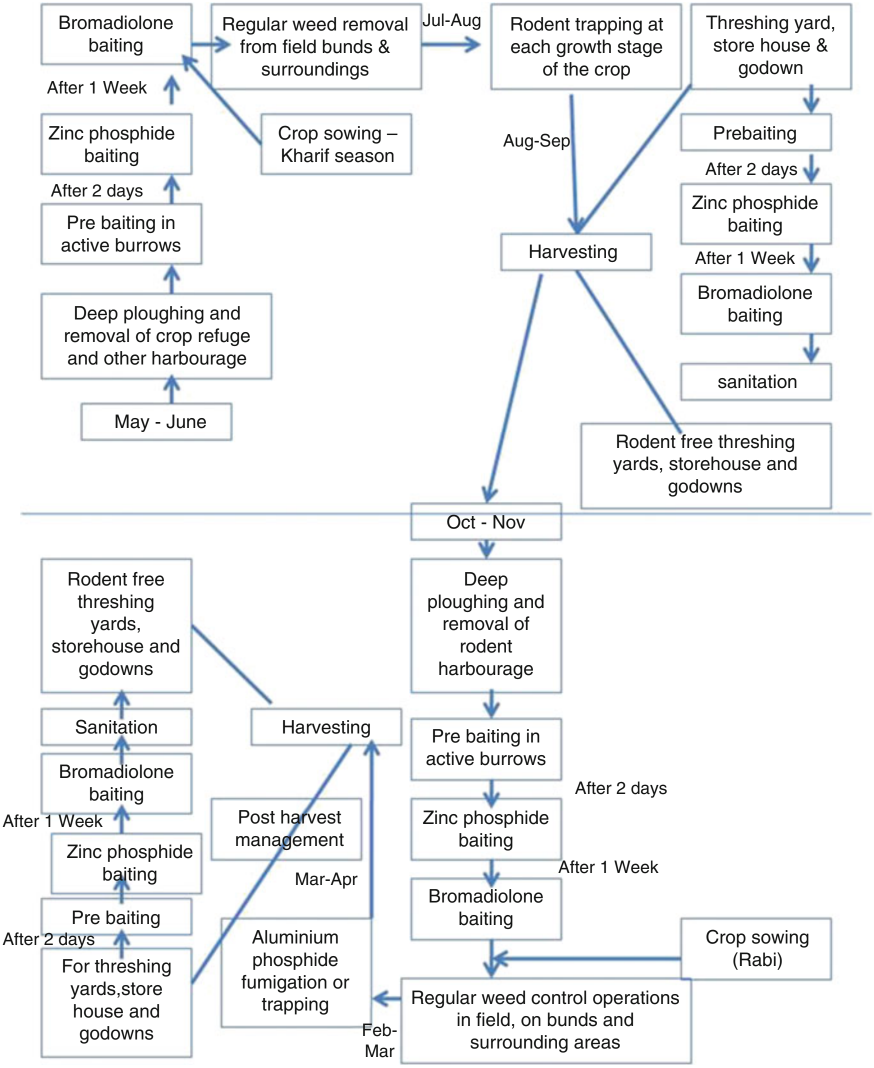


Fig. 11.6 Integrated management schedule for pest rodents

and postcontrol figures gives per cent success of the operation. In store/warehouses, godowns and poultry units, the food consumption index during precontrol and postcontrol censuses would give the control success.

11.9.4 Monitoring and Review

Monitoring and review are the essential component of rodent control programme with the following essential data:

- Personnel responsible for rodent control programme and place of the programme
- Environmental risk analysis
- Rodenticide used in the campaign
- Bait placement map
- Quantity of rodenticide used
- Important dates of all operations
- Bait consumed and bait station used
- Number of dead rodents
- Records of monitoring
- Details on leftover baits
- Rodent disposal
- Completion of the campaign

All these data should be analysed so that progress of the control programme can be easily known.

11.10 Conclusion

Rodents pose major threats to crop production, sometimes strikingly obvious, sometimes more cryptic. Awareness of their biology can improve the prospects for their management. Since rodents are highly evolved mammals, they avoid trouble by trap and poison avoidance, bait shyness/aversions to acute poisons, resistance or cross-resistance to rodenticide, etc. Therefore, an integrated approach based on the biology, ecology and ethology of the target rodent pest species *vis-a-vis* control measures which are cost-effective and socially acceptable is to be evolved. Sound knowledge on population ecology of the species concerned, behaviour of the pest, damage threshold of the pest on crops, state of dominance of the pest, human interference, etc., plays a vital role in the successful planning and implementation of rodent management programme, so that the outcome is more sustainable with more favourable economics. Therefore, understanding their bioecology is the prerequisite to have a long-term and sustainable rodent pest management programme.

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