Soil Health Indicators: Methods and Applications

13

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Abstract

Soil health indicators are a composite set of measurable physical, chemical, and biological attributes which relate to functional soil processes and are being used to evaluate soil health status. A range of soil health indicators have been developed to measure and asses changes in soil properties and functioning to understand soil health as a tool for sustainability. The physical, chemical, and biological indicators must be employed to verify soil status use and to undertake remedial management measures within a desired timescale. Soil properties which can change rapidly in response to natural or anthropogenic actions are considered as good soil health indicators. Among the physical indicators, bulk density, soil aggregate stability, and water holding capacity have been found ideal indicators. Chemical indicators such as pH, EC, soil organic carbon, and soil nutrient status are well established. However, most of them generally have a slow response, as compared to the microbiological and biochemical properties, such as soil enzymes, soil respiration, mycorrhiza, lipid profiling, and earthworms as they change rapidly due to perturbation caused by different agricultural management paradigm. Thus, systemic approaches based on different kinds of indicators (physical, chemical, and biological) in assessing soil health are discussed in this chapter.

Keywords

AMF · Earthworm · Enzymes · PLFA · Soil health indicators · Soil health management · Soil quality

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13.1 Introduction

Modern agricultural practices began to exploit soil by excessive use of chemical fertilizers devoid of organic sources, nature of the soil such as high pH, $CaCO_3$ and low organic carbon content; extensive tillage with heavy machinery and closely spaced cereal–cereal rotations. This has instead of created insert caused multifaceted deleterious effect on soil health by reducing time required by the soil health indicators (biological) to rejuvenate and perpetuate for maintaining ideal environment condition for crop growth without compromising on economic yield. Moreover, this situation has accelerated soil degradation process insidiously making roads into weakening of soil health indicators to become unproductive soil (Katyal et al. 2016). At present, demand for sustainable agricultural management practices mounting due to agricultural edges has already expanded near to the maximum all over the world. Feeding ever increasing population with maintaining optimum soil health indicators and sustainable environment is ever challenging task for present and future generations to come. In addition, public awareness and thrust on the need of environmental conservation, especially in the tropical region, claim for keeping forests as reserve of biodiversity, provider of environmental services, and needs for reclamation of degraded lands (Cardoso et al. 2013) is also a matter of great concerns. Therefore, sustainable agricultural practices to maintain optimum soil heath indicators with ideal soil fertility are needed for meeting the needs of the present without compromising the productive potential for the next generations. The rational soil use practices must allow economically and environmentally sustainable yields, and also quality of produce which will only be reached with the maintenance or recovery of the soil health indicators. Thus, a healthy soil has "the continued capacity of soil to function as a vital living system, within ecosystem and land-use boundaries, to sustain biological productivity, promote the quality of air and water environments, and maintain plant, animal and human health" (Doran and Safley 1997). To assess the sustainability of a production system, changes in soil health indicators (chemical, physical, and biological) and their effects on the soil's capacity to support plant growth and external environment functions must be monitored. Hence, in this chapter an impetus has been given to discuss soil health indicators in detail with methodologies to analyze them in the laboratory along with their potential applications in crop production and management aspects under field conditions.

13.2 General View of Soil Health Indicators

The soil consists of four major components such as air, water, mineral, and organic matter that are described in terms of soil health indicators, which can provide an assessment of how well the soil functions. Though the properties that constitute a healthy soil are not the same in all situations and locations, there are some important soil properties that indicate soil health. Soil health indicators are selected based on soil characteristics, soil use, and environmental circumstances along with their positive correlation with crop growth and yield under different management

Table 13.1 Soil health indicators selected based on certain criteria (modified from Arshad and Coen 1992; Idowu et al. 2008; Kelly et al. 1999; Paoletti et al. 2010; Griffiths et al. 2018)

Soil health indicators	Rationale for selection
Bulk density	Plant root penetration, porosity, adjust analysis to volumetric basis
Soil aggregate stability	Soil structure, erosion resistance, crop emergence an early indicator of soil management effect
Water holding capacity/infiltration	Runoff, leaching, and erosion potential
pH	Nutrient availability, pesticide absorption and mobility, process models
EC (electric conductivity)	Defines crop growth, soil structure, water infiltration; presently lacking in most process models
CEC (cation exchange capacity)	CEC represents the total amount of exchangeable cations that soil can absorb
Soil organic carbon/organic matter	Defines soil fertility and soil structure, pesticide and water retention, and use in process models
Soil nutrients status	Availability of crops, leaching potential, mineralization/immobilization rates, process modeling, capacity to support plant growth, environmental quality indicator
Suspected pollutants	Plant quality, and human and animal health
Soil respiration	Biological activity, process modeling; estimate of biomass activity, early warning of management effect on organic matter
Enzymes (dehydrogenase, β-glucosidase, acid and alkaline phosphatase, microbial biomass, and soil respiration)	Electron transferences in the respiratory chain in living cells, C oxidation, organic phosphorus cycling, source and/or drain of C and nutrients, microbial mineralization of organic carbon
Mycorrhiza	Nutrient mobilization, soil aggregation
Trichoderma	Residue decomposition
Lipid profiling	Diversity and biomass
Earthworm	Indicate relative change in soil structure, nutrient recycling, regulate soil water, aeration, and provide drainage

conditions (Cardoso et al. 2013). Some of the key soil health indicators for soil quality assessment are provided in Table 13.1 and the inter relationship between different soil health indicators are emphasized in Table 13.2. According to Bünemann et al. (2018), the most commonly used and frequently proposed soil health indicators by various authors across the globe are soil organic carbon and soil pH (Fig. 13.1), followed by available phosphorus, indicators of water storage, and bulk density. The soil texture, available potassium, and total nitrogen are also frequently used (>40%). For soil reclamation point of view, the important soil properties that indicate soil health could be physical, chemical, biological, or

Table 13.2 Interrelationship of soil indicators (Laishram et al. 2012)

Selected indicator	Other soil quality indicators
Aggregation	Organic matter, microbial (especially, fungal) activity, texture
Water holding capacity/ infiltration	Organic matter, aggregation, electrical conductivity, exchangeable sodium percentage (ESP)
Bulk density	Organic matter, aggregation, topsoil-depth, ESP, biological activity
Microbial biomass	Organic matter, aggregation, bulk density, pH, texture, ESP, and/or respiration
Available nutrients	Organic matter, pH, topsoil-depth, texture, microbial parameters (mineralization and immobilization rates)

biochemical within that average number of indicators selected based on their practical and economical feasibility as well as their relations with other indicators under all the conditions are described in this chapter.

13.3 Soil Health Indicators and Their Analytical Techniques

13.3.1 Soil Physical Health Indicators

Soil physical health indicators provide information related to water and air movement through soil, as well as conditions affecting germination, root growth, and erosion processes. Thus, soil physical health indicators form the foundation for other chemical and biological processes. Key soil physical indicators in relation to crop production include soil aggregate stability, water holding capacity, bulk density and are discussed below.

13.3.1.1 Water Holding Capacity and Bulk Density

Soil water holding capacity is the amount of water a given soil can hold for crop use. How much water a soil can hold is very important for crop production point of view. Soils which hold more water can support higher plant growth and development and reduce leaching losses of nutrients and pesticides. Hence, water holding capacity of soils is explained in terms of infiltration, soil available water and distribution. Soil water infiltration, the rate at which water enters the soil surface and moves through soil depth, is gaining increased interest (Dalal and Moloney 2000; Joel and Messing 2001). Since infiltration rate may change significantly with soil use, management, and time, it has been included as an indicator of soil health for assessments of land use change impacts (Arias et al. 2005; O'Farrell et al. 2010).

Bulk density is the weight of dry soil per unit of volume expressed in grams cm^{-3}. It is routinely assessed in agricultural systems to characterize the state of soil compactness in response to land use and management (Håkansson and Lipiec 2000). It has been considered as a useful indicator for the assessment of soil health with respect to soil functions such as aeration, infiltration (Reynolds et al. 2009), rooting depth/restrictions, available water capacity, soil porosity, plant nutrient availability, and soil microorganism activities influencing the key soil processes and productivity

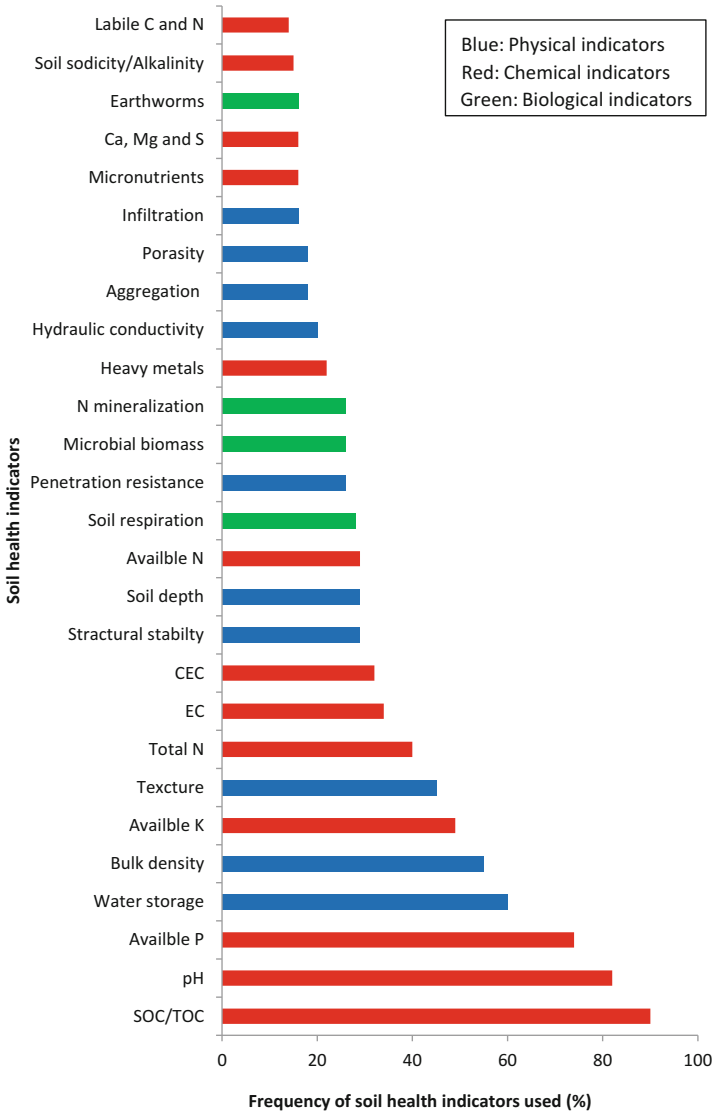

Fig. 13.1 Frequency of different indicators used all over the world (Modified from Bünemann et al. 2018)

(https://www.nrcs.usda.gov). Since bulk density in general is negatively correlated with soil organic matter (SOM) or SOC content (Weil and Magdoff 2004), loss of organic C from increased decomposition due to elevated temperatures (Davidson and Janssens 2006) may lead to increase in bulk density and hence making soil more prone to compaction through land management activities (Birkas et al. 2009). Bulk density directly measures compaction, and generally does not vary with other soil

properties because it is most often expressed on a dry soil basis (Tokunaga 2006). It has been suggested by many researchers that soil bulk density from 1.3 to 1.7 mg m^{-3} may limit root growth and decrease plant yield (Asady and Smucker 1989; Bengough and Mullins 1990; Kuznetsova 1990). Maximum water holding capacity of soil was assessed with Keen Raczkowski cup as per the method described by Piper (1966). Bulk density of soil sample is determined by using core sampler technique (Black 1965), recording the fresh weight of the sample in the field and dry weight of the sample in the laboratory. Drying of soil can be done in hot air oven to constant weight. Bulk density calculation was done as dry weight of soil per unit volume of the core collect with core sampler in the field. The units are expressed as % and g cm^{-3} for water holding capacity and bulk density, respectively.

13.3.1.2 Aggregate Stability

Aggregate stability is an indicator of organic matter content, biological activity, and nutrient cycling in soil and is determined by soil structure as influenced by a range of chemical and biological properties and management practices (Dalal and Moloney 2000; Moebius et al. 2007). It is considered as a useful soil health indicator since it is involved in maintaining important ecosystem functions in soil including organic carbon (C) accumulation, infiltration capacity, movement and storage of water, and root and microbial community activity; it can also be used to measure soil resistance to erosion and management changes (Moebius et al. 2007; Rimal and Lal 2009). Aggregate stability is crucial for soil health which can be measured with the methods proposed by Kemper and Chepil (1965) (a dry sieving and wet sieving), Bissonnais (1996) and Six et al. (2000) (the method does not require the use of equipment to mechanically submerge sieves, pre-sieving dry aggregates but rather is done by hand). The most common method used for aggregate stability measurement is wet sieving (Haynes 1993). The disadvantage of the method proposed by Bissonnais (1996) is that aggregate stability is increased by sand particles that are not excluded from the calculation of coefficient of vulnerability (Kv). On the other hand, a big advantage of this method is distinguishing the particular mechanisms of aggregate breakdown. Therefore, it can be used within a large range of soils. In the assessment of water stable aggregate (WSA), only hexa-metaphosphate as a dispersing solution was used, because sodium hydroxide was too aggressive to the aluminum cans. An advantage of this method is that sand particles are excluded from the calculation of WSA index.

13.3.2 Soil Chemical Health Indicators and Their Analytical Techniques

Soil chemical health indicators are correlated with the capacity to provide nutrients for plants and/or retaining chemical elements or compounds harmful to the environment and plant growth. Soil pH, electrical conductivity, cation exchange capacity (CEC), soil organic carbon, and nutrient status are the main chemical indicators used

in soil health assessment, especially when considering the soil capacity for supporting high yield crops (Kelly et al. 1999).

13.3.2.1 Soil pH, Electrical Conductivity, and Cation Exchange Capacity

Soil pH is one of the most indicative measurements of the chemical properties of soil. Whether a soil is acidic, neutral, or basic has much to do with solubility of various compounds, the relative bonding of ions to exchange sites, and the various microorganisms. Soil pH can be determined by an electrometric method (Jackson 1973) using a glass electrode pH meter in a 1:2 suspension of soil and water by using buffer solutions at pH 4.0 and 7.0, the pH read on pH meter. Soil electrical conductivity (EC), a measure of salt concentration, is considered an easily measured, reliable indicator of soil quality/health (Arnold et al. 2005). It can inform trends in salinity, crop performance, nutrient cycling (particularly nitrate), and biological activity and, along with pH, can act as a surrogate measure of soil structural decline especially in sodic soils (Dalal and Moloney 2000; Arnold et al. 2005). Electrical conductivity has been used as a chemical indicator to indicate soil biological quality in response to crop management practices (Vargas Gil et al. 2009). Clearly, there is a need for a comprehensive assessment of soil EC as an important soil health indicator in different ecosystems (Smith et al. 2002). Electrical conductivity of soil samples can be determined by the method suggested by Piper (1966) using a conductivity meter (Chemita 130) in 1:2 (soil:water ratio).

Cation exchange capacity (CEC) is also considered as an important determinant of soil chemical quality, particularly the retention of major nutrient cations Ca, Mg, and K and immobilization of potentially toxic cations Al and Mn; these properties can thus be useful indicators of soil health, informing of a soil's capacity to absorb nutrients, as well as pesticides and chemicals (Dalal and Moloney 2000; Ross et al. 2008). Ion exchange capacity mostly affects soil cation exchange capacity (CEC) binding to negative charge organic matter, clay, and soil colloid. CEC in soil can be measured by ammonium acetate method (Schollenberger and Dreibelbis 1930) at pH 7 and the barium chloride-triethanolamine method (Mehlich 1938) at pH 8.2.

13.3.2.2 Soil Organic Carbon

Soil organic carbon is a key attribute in assessing soil health, generally correlating positively with crop yield (Bennett et al. 2010). The soil organic carbon affects important functional processes in soil like the storage of nutrients, mainly N, water holding capacity, and stability of aggregates (Silva and SáMendonça 2007). In addition, the soil organic carbon also affects microbial activity. Hence, this is a key component of soil fertility, especially in tropical conditions, which interacts with chemical, physical, and biological soil properties and must be considered in assessments of soil health. Soil organic carbon content can be measured with help of Walkley and Black method. The method involves the oxidation of potassium dichromate solution in sulfuric acid medium and evaluating the excess of dichromate with titration against ferrous ammonium sulfate (Yeomans and Bremner 1988). Weil et al. (2003) reported a highly simplified method using slightly alkaline $KMnO_4$ to analyze oxidizable (active) forms of soil C. They showed that the active soil C

measured was more sensitive to soil management practices than total organic C, and more closely related to biologically mediated soil properties, such as respiration, microbial biomass, and aggregation, than several other measures of soil organic C.

13.3.2.3 Available Nutrients (N, P, S, Zn, and Fe)

Available soil nutrients (N, P, K, S, Zn, and Fe) and their identification of basic soil properties to meet requirements of indicators for screening soil health, Doran and Safley (1997) proposed extractable nutrients as "they provide information on plant available nutrients and potential loss from soil providing indication of productivity and environment quality." Measurement of extractable nutrients may provide indication of a soil's capacity to support plant growth; conversely, it may identify critical or threshold values for environmental hazard assessment (Dalal and Moloney 2000). Nutrient cycling, especially N, is intimately linked with soil organic C cycling (Weil and Magdoff 2004) and possibly the cycling of other plant available nutrients. The mineralizable nitrogen in soil can be determined with help of alkaline permanganate method (Subbiah and Asija, 1956) using a Kjeldahl distillation unit. The available phosphorous can be extracted with Olsen's reagent (0.5M $NaHCO_3$, pH 8.5) in neutral to alkaline soils (Olsen et al. 1954), whereas under acid soils Brays P-1 (0.03N NH_4 F and 0.025N HCL) is widely followed (Bray and Kurtz 1945). The major drawback with blue color development (Dickman and Bray 1940) is that color starts fading soon and hence intensity has to be measured quickly. Therefore ascorbic acid method (Watanabe and Olsen 1965) provides stable blue color and therefore preferred over former methods to estimate available phosphorus in soil. Available sulfur can be extracted by using Morgan's universal extractant (pH 4.8) and is determined by turbidimetric method (Chesnin and Yien 1950) using UV/Visible spectrophotometer. For micronutrients extraction, neutral ammonium acetate and chelating agents like EDTA and DTPA have been used for extraction of Zn, Fe, Cu, and Mn from soil and the extracted amount is determined calorimetrically. Zn determination dithizone method (Shaw and Dean 1952) has been very popular until AAS become available. For those laboratories where AAS is not yet available the alternative (colorimetric) methods as described by Jackson (1973) are still employed. However, for rapid and accurate analysis of Zn, Fe, Cu, and Mn the DTPA method (Lindsay and Norvell 2010) is most widely used to estimate micronutrients.

13.3.3 Microbiological and Biochemical Health Indicators and Their Analytical Techniques

Soil microbial activity and diversity play an important role in the sustainability by keeping essential functions of soil health, involving carbon and nutrient cycling (Jeffries et al. 2003; Izquierdo et al. 2005). Microbial indicators are more sensitive than physical and chemical attributes to changes imposed to the environment like soil use and management (Masto et al. 2009). Some of the commonly used soil

biochemical/biological parameters which depict the soil quality status of a given soil along with their analytical techniques are illustrated below:

13.3.3.1 Soil Microbial Biomass (Microbial Biomass Carbon (MBC) and Microbial Biomass Nitrogen (MBN))

The soil microbial biomass (MBC and MBN) is the active component of the soil organic pool and plays an important role in nutrient cycling, plant nutrition, and functioning of different ecosystems. It is responsible for organic matter decomposition thus affecting soil nutrient content and, consequently, primary productivity in most biogeochemical processes in terrestrial ecosystems (Gregorich et al. 2000; Haney et al. 2001). In the last 30 years, relatively rapid assessment of soil microbial biomass has been possible based on physiological, biochemical, and chemical techniques (Horwath and Paul 1994) such as chloroform fumigation incubation (CFI) (Jenkinson and Powlson 1976), chloroform fumigation extraction (CFE) (Brookes et al. 1985; Vance et al. 1987), substrate-induced respiration (SIR) (Anderson and Domsch 1978), and adenosine triphosphate (ATP) analysis (Jenkinson et al. 1979; Eiland 1983; Webster et al. 1984). Microbial biomass has even been proposed as a sensitive indicator of soil quality (Karlen et al. 1997) and soil health (Sparling 1997). Of these, the first two methods have been widely used to estimate microbial biomass in agricultural, pastoral, and forestry systems, rehabilitation of disturbed lands, and pesticide and heavy metals polluted soils. The methods are used to analyze microbial biomass carbon and nitrogen as explained in detail below.

Chloroform Fumigation Incubation (CFI)

In this method, a moist soil is fumigated with ethanol free chloroform for 24 h; chloroform is then removed by repeated evacuation; the soil is reinoculated with a small amount of unfumigated soil and then incubated at a constant temperature (usually 22 or 25 °C) for 10 days at field capacity or 50% of its water holding capacity (about −0.01 MPa). An additional soil sample is retained unfumigated and used as a control. The CO_2 evolved during incubation can be measured by gas chromatography, as a continuous flow or by sorption in alkali followed by titrimetric, conductometric, or colorimetric determination. As the net C mineralized as CO_2 is only a proportion of the total microbial biomass C, a *k*C factor is used to calculate total soil biomass C. As for as soil microbial biomass N determination, mineral N (NH_4-N and NO_3-N) from both fumigated and unfumigated (control) samples are extracted with 2Ml KCl after incubation. The mineral N in the extracts is then determined colorimetrically or by steam distillation. As for microbial biomass N, a *k*N factor is used to correct for incomplete mineralization of N from killed microorganisms for calculating total biomass N. Soil microbial biomass C and N are calculated from equations (1) and (2): Biomass C = (CO_2-C fumigated − CO_2-C control)/*k*C (1), Biomass N = (mineral N fumigated − mineral N control)/*k*N (2). The widely accepted *k*C value is 0.41 at 22 °C (Anderson and Domsch 1978) or 0.45 at 25 °C (Jenkinson and Powlson 1976). However, *k*N varies from 0.30 to 0.68 (Smith and Paul 1990). Jenkinson (1988) suggested a *k*N value of 0.57 at 25 °C,

which is about 0.50 at 22 °C. Two basic assumptions of the CFI method are: (1) that CO_2-C evolved or mineral N produced during incubation in fumigated soil must exceed that from the corresponding unfumigated soil; and (2) that CO_2-C evolved or mineral N produced during incubation from the non-microbial source must be equal in both fumigated and unfumigated soil samples (Jenkinson 1988). In soils with relatively low microbial biomass but high respiration activity, subtraction of the CO_2 evolved from an unfumigated sample (control) often leads to low or even negative biomass estimates because unequal amounts of non-microbial biomass C is mineralized (Horwath et al. 1996). To overcome this problem, Jenkinson and Powlson (1976) suggested that CO_2-C released during the 10–20 day incubation rather than that from the initial 0–10 day incubation of unfumigated soil should be subtracted from the CO_2-C released from the fumigated soil. Horwath et al. (1996) suggested that the proportion of CO_2-C subtracted from the unfumigated (0–10 day incubation) soil should vary as a function of the ratio of CO_2-C fumigated/CO_2 control. When the ratio is large the proportion of CO_2-C subtracted from the unfumigated soil should be large and vice versa. They also suggested that equation (1) can be modified to: Biomass C = (0.71 × CO2-C fumigated – 0.23 × CO_2-C controls)/*k*C. However, the modified equation needs to be validated for soils under different land use and management and in different climates. The two basic assumptions mentioned above do not hold for soils with pH <5, air-dried soils, waterlogged soils, and soils that contain recently added organic materials or plant residues. In acidic soils, the re-establishment of a C and N mineralizing microbial population after fumigation and reinoculation is very slow. This causes a reduced mineralization of the killed microorganisms which makes the usual *k*C and *k*N factors invalid (Jenkinson 1988; Martens 1995). In air-dried soils, the amount of already dead microorganisms may constitute most of the microbial biomass in both fumigated and unfumigated soil samples, in addition to the less effective lysing of microbial cells by chloroform (Sparling and West 1989). In waterlogged soils, CO_2 and CH_4 are produced under conditions that restrict diffusion of gases (Jenkinson 1988). In soils with recently added organic materials or plant residues, the second assumption is not met since the mass of the re-established microbial population in the fumigated and reinoculated soil sample corresponds to only 10–20% of the original microbial biomass and consists mainly of bacteria. This can be avoided by either careful removal of the amendments such as roots, or a sufficient preincubation of at least 3 weeks (Martens 1995).

Chloroform Fumigation Extraction (CFE)

The above-mentioned limitations of the CFI method are mainly overcome by extraction of C and N with 0.5 mol K_2SO_4/L from the chloroform fumigated and the unfumigated soil samples. The proportions of C (*k*EC) and N (*k*EN) extracted from the fumigated (killed microbial biomass) soil vary from 0.2 to 0.68 (Jenkinson 1988; Martens 1995). However, most frequently used *k*EC values are in the range 0.36–0.45, while the *k*EN values are in the range 0.49–0.62. Likely limitations of the CFE method are differential extraction of released C from soils that differ in clay content and clay mineralogy, and variable *k* values (Martens 1995). The CFE method

has been successfully used to estimate soil microbial biomass P (Hedley and Stewart 1982) and S (Saggar et al. 1981). Inorganic P is extracted with 0.5 mol Na_2HCO_3/L (pH 8.5) from both a fumigated and an unfumigated soil; the proportion of P is extracted from the killed microbial biomass, and the *k*P value is taken as 0.4. The allowance is also made for P sorption during fumigation and extraction by including an internal P standard. For strong P retention soils such as Ferrosols, Bray extractant (30 mmol NH_4F/L + 25 mmol HCl/L) appears to be more appropriate than 0.5 mol Na_2HCO_3/L extractant (Oberson et al. 1997). The procedure for microbial biomass S determination is similar to that for microbial biomass P but 0.15% $CaCl_2$ is used as an extractant and determined using turbidimetric method, the most commonly used *k*S value is 0.41 (Smith and Paul 1990).

Substrate-Induced Respiration (SIR)

An excess of substrate, usually glucose, is added to a soil, which is then incubated at constant temperature and moisture, and the respiration rate, CO_2 evolved per hour, is measured during a 0.5–2.5 h period, before the microorganisms start proliferating and actually increase microbial biomass (Anderson and Domsch 1978). Limitations of this method are: (1) that the pattern of soil microbial response to glucose differs between soils; (2) that only glucose responsive soil microbial biomass is measured; (3) that soils recently amended with organic materials or plant residues contain a large proportion of young cells, and, therefore, the conversion factor used, from mL CO_2/h to microbial biomass C of 40 (30 at 22 °C, Beck et al. 1997) for an average population in soil, is not valid (Martens 1995); (4) it measures only microbial activity which does not necessarily equate with microbial biomass; and (5) that microbial biomass N, P, and S cannot be measured (Smith and Paul 1990).

Adenosine Triphosphate Analysis (ATP)

Adenosine triphosphate is a universal constituent of living microbial cells. Although ATP can occur in dead microbial cells and extracellularly in soil, it is rapidly degraded by microorganisms. Therefore, ATP concentration in soil can be used to estimate the amount of living microbial biomass. It is usually extracted with acid reagents from moist, preincubated soil, and estimated by the luciferin–luciferase system. The C: ATP ratio is about 200 although it varies from 120 to 240 (Jenkinson et al. 1979; Eiland 1983; Martens 1995). The limitations of the ATP method are: (1) that ATP is decomposed by enzymatic and chemical hydrolysis during the extraction process; (2) after its release from microbial cells, ATP is strongly sorbed by soil constituents (Martens 1995); (3) biomass C: ATP ratio changes substantially over time in response to soil amendments such as organic materials and plant residues (Tsai et al. 1997); and (4) it cannot measure microbial biomass N, P, and S in soil (Smith and Paul 1990).

Phospholipid Fatty Acids

Phospholipid fatty acids with a chain length of $<$20 C atoms are considered to be of mainly bacterial origin (Harwood and Russel 1984). However, 18-C chain phospholipid fatty acid, 18: 2ω6 fatty acid constitute on average 43% of the total

phospholipid fatty acid in soil fungi (Federle et al. 2010). Since ergosterol is specific to the fungal membrane (Seitz et al. 1979), the fungal biomass can be estimated from the correlation between the amounts of 18:2ω6 fatty acid and the ergosterol content. Frostegard and Baath (1996) observed a close correlation between the amounts of 18:2ω6 fatty acid and the ergosterol in soil ($r = 0.92$), thus, indicating that this phospholipid fatty acid can be used to estimate fungal biomass. The ratio of 18: 2ω6 fatty acid:bacterial phospholipid fatty acids is then used as a fungal:bacterial biomass ratio (Frostegard and Baath 1996). Phospholipid fatty acids can be extracted from soil with a one-phase mixture of chloroform, methanol, and citric acid buffer, fractionated into neutral, glyco- and phospholipids on columns containing silicic acid, methylated into fatty acid methyl esters, and then measured on a gas chromatograph/mass spectrometer. The advantage of the phospholipid fatty acid method, compared with other methods to estimate the microbial biomass of individual communities, is that both fungal and bacterial biomass can be estimated by the same technique in a single soil extract (Frostegard and Baath 1996). Currently PLFA analysis in soil and roots are being analysed using high throughput method, where PLFA is being eluted through 5:5:1 (chloroform, methanol, water) through column chromatography and eluted PLFA were transesterified and FAME profiles were identified using the MIDI PLFAD1 calibration mix and peak naming table through MIDI (MIDI, Inc., Newark, DE) system attached with GC (Buyer and Sasser 2012; Sharma and Buyer 2015). Although high throughput method is rapid, cost effective, and has added technical advantages than conventional method. However, its uses are limited due to high instrumentation costs and technical skills.

Ninhydrin Reaction Method

Amato and Ladd (1988) proposed to use ninhydrin reactive C and N compounds released during fumigation incubation as a measure of biomass. They specifically determined that fumigated soils retained protease but lost dehydrogenase activity required to decompose glucose and immobilize NH_4-N during the incubation period. They proposed to quantify ninhydrin reactive N compounds released in CFI (10 days incubation at 25 °C, extraction with 2N KCl) and determine biomass N by using a multiplication factor of 21. Thus the method differs from original CFI in which ninhydrin reactive C and N compounds rather than NH_4-N (or total mineral N) and CO_2 are taken into consideration while calculating biomass. Ocio and Brookes (1990) considered the ninhydrin method suitable for freshly amended soils (CFI gives unreliable results for such soils) and found good correlation with CFE and SIR. Sparling (1997) concluded that the ninhydrin method can give a reliable estimate of biomass in organic as well as mineral soils. Van Gestel et al. (1993) also determined biomass C indirectly by multiplying ninhydrin reactive extractable N of fumigated soils with 21 (Amato and Ladd 1988); they used 2N KCl for extraction. As compared with original CFI, the ninhydrin reaction method is less preferred due to its long processing time (at least 10 days is required for obtaining biomass values), nevertheless it has advantages due to its reliability in results particularly for freshly amended soils or soils rich in easily oxidizable C.

Microcalorimetry

Sparling (1981) proposed microcalorimetry as a method to assess microbial metabolism in soil on the pretext that the heat produced depends only on the initial and final energy states of the system and is independent of the types of organisms or reaction pathway. In addition, the total catabolic activity in the soil is closely related to the heat production; anabolic processes normally contribute a little to the heat. Sparling (1981) studied heat output from 12 soils and compared the results with CFI and SIR, ATP, dehydrogenase and amylase, and basal respiration. The rate of heat output from soil is closely related with the rate of respiration. Heat is found to be less correlated with most of parameters used. Hence, microcalorimetry method has not achieved popularity to a significant extent.

Microwave Irradiation

Microwave irradiation is an effective biocide treatment of soil which kills weeds, nematodes, and microorganisms; the effect on microorganisms being probably entirely thermal (Vela and Wu 1979), fungi being more susceptible (Wainwright et al. 1980). Spier et al. (1986) were probably the first to use microwave radiation for soil treatment to measure microbial biomass, an approach akin to $CHCl_3$ fumigation. In spite of its simplicity, this method has not gained widespread acceptability.

13.3.4 Comparison of Different Methods to Estimate Soil Microbial Biomass

Currently, all methods used to analyze soil microbial biomass have some limitations since these were developed for soils with microbial biomass in a relatively steady state. The soil microbial biomass has been measured through various methods in which values are variable due to having different k factors, soils at different moisture contents, different incubation temperatures, soils containing variable amounts of organic materials or plant residues, and different instrumentation and analytical techniques. Therefore, it is difficult to compare and get reproducible soil microbial biomass values obtained by different methods in different laboratories (Dalal 1998; Azam et al. 2003).

13.3.5 Soil Enzymes

Soil enzymes play a key role in the energy transfer through decomposition of soil organic matter and nutrient cycling, and hence play an important role in agriculture. Soil enzymes, being necessary catalysts for organic matter recycling, strongly influence on soil fertility and agronomic productivity (Rao et al. 2014). Soil enzymes are highly sensitive and quickly respond to any changes in soil management practices and environmental conditions. Their activities are closely related to physio-chemical and biological properties of the soil. Hence, soil enzymes are used as sensors for soil microbial status, for soil physio-chemical conditions, and

for the influence of soil treatments or climatic factors on soil fertility. Understanding the possible roles of different soil enzymes in maintaining soil health can help in the soil health and fertility management, particularly in agricultural ecosystems (Rao et al. 2017). Some of the frequently analyzed soil enzymes for soil health point of view are discussed.

Phosphomonoesterase, i.e., acid and alkaline phosphatase activity in rhizosphere soil sample is determined using the procedure of Tabatabai (1994) with the following modification as suggested by Schinner et al. (1996). Arylsulfatase activity is measure by adopting the method of Sarathchandra and Perrott (1981). ß-Glucosidase is determined using *p*-nitrophenyl-ß-D-glucopyranoside (PNG, 0.05M) as substrate. This assay is based on the release and detection of p-nitrophenol (PNP) (Tabatabai 1982). Dehydrogenase activity is measure with reduction of 2,3,5-triphenyl-tetrazolium chloride (TTC) to triphenyl formazan (TPF) using colorimetric procedure of Tabatabai (1994). Fluorescein diacetate (FDA) hydrolysis is determined by the method of Schnürer and Rosswall (1982) and Aseri and Tarafdar (2006). Urease activity (urea amidohydrolase) is determined by the non-buffer method of Zantua and Bremner (1975).

13.3.6 Arbuscular Mycorrhizal Fungi

Arbuscular mycorrhizal fungi (AMF) establish a symbiotic relationship with more than 80% of terrestrial plants (Brundrett 2002). In order to establish a new mycorrhizal association, AMF forms infectious propagules such as spores, extraradical phase consisting of hyphae that develops into the soil, and intraradical phase consisting of arbuscules and vesicles (Linderman 1997) where its colonizing ability varies from species to species (Klironomos and Hart 2002). Spores proved efficient for infecting roots for *Gigaspora* and *Scutellospora* species whereas for *Glomus* and *Acaulospora* all inoculum forms were found to be equally efficient (Klironomos and Hart 2002). Several factors come into play while shaping the AMF community composition such as agricultural management practice (Jansa et al. 2006; Oehl et al. 2010; Curaqueo et al. 2011); soil type (Oehl et al. 2010); and concentration of nutrients (Gosling et al. 2013) and host species (Lovelock et al. 2003; Gosling et al. 2013), etc. AMF draws nutrients from the soil with the help of its extraradical hyphae for the use of the plant and receives photosynthates from plant in the root cortex as well as in the rhizospheric region (Smith and Read 2008). AMF together with fibrous roots facilitates the formation of sticky string bag where it mechanically binds soil aggregates together forming macroaggregates (Miller and Jastrow 2000). Practices such as tillage cause the mechanical disruption of hyphae (Boddington and Dodd 2000). AMF has also been credited with the production of heat-stable glycoprotein called glomalin (Wright and Upadhyaya 1996). Glomalin acts a soil particle cementing agent and its concentration strongly relates with soil aggregate stability (Wright and Upadhyaya 1998). Hence AMF are integral component of plant rhizosphere where array of microbial activities are taking place. The stabilized crop and soil conservation practices enhance AMF biomass (Sharma et al. 2012). Therefore

AMF can be used as potential indicator to assess the sustainability of long-term farming systems. The AMF biomass can be determined through microscopic and biochemical methods in terms of spore's density (Gerdemann and Nicolson 1963), root colonization (Phillips and Hayman 1970), and 16:1ω5cis PLFA and NLFA as AMF signature fatty acids (Sharma and Buyer 2015; Olsson 1999). Signature fatty acid analysis provides a more promising approach over the conventional methods. Glomalin has also been used as an indicator of AMF (Krivtsov et al. 2004). In the following sections we have provided a comprehensive assessment of techniques used for the quantification of AMF biomass. Quantification of AMF biomass has mainly been done through microscopic methods (Gerdemann and Nicolson 1963; Phillips and Hayman 1970).

13.3.6.1 Microscopic Methods of AMF Quantification

The quantification of AMF biomass is performed conventionally through extracting spores by wet sieving and decantation method (Gerdemann and Nicolson 1963). The suspension obtained can be observed directly or filtered through a filter paper disc and spores are counted under a microscope. For the assessment of root colonized by AMF, the techniques used include the root staining (Phillips and Hayman 1970) followed by quantification using the gridline intersect method (Giovannetti and Mosse 1980) that provides an estimate of root length colonized by AMF. Other important parameters include the measurement of hyphal dry weight and microscopic examination of stained hyphae for the study of extraradical hyphal length and hyphal connections (Miller et al. 1995; Mosse 2009).

13.3.6.2 Signature Fatty Acid Analysis

The intensity of response unveiled by the membrane lipids to instabilities/disturbances is highest (Denich et al. 2003). For the quantification of AMF signature fatty acid PLFA 16:1ω5cis has been extensively used (Olsson et al. 1995). Phospholipid 16:1ω5cis is a reflection of AMF extraradical hypha length and neutral lipid 16:1ω5cis portrays storage lipids that include spore copiousness (Olsson et al. 1997). Ester-linked fatty acids (ELFAs) include all the three major classes of lipids such as phospholipid, neutral lipid, and glycolipids (Sharma and Buyer 2015). ELFA 16:1ω5cis and 18:1ω5cis have also been used to study AMF dynamics (Grigera et al. 2007). Lipids are extracted through the Bligh–Dyer extraction method (Bligh and Dyer 1959) which is followed by division of lipids into phospholipids, neutral lipids, and glycolipids, which are later exposed to mild alkaline methanolysis and analyzed on a gas chromatograph (Frostegard et al. 1993). The use of solid phase extraction (SPE) technique by means of column chromatography further improves the extraction efficiency (Zelles et al. 1992; Zelles 1999). To advance further, a high throughput method was introduced that permitted the analysis of a batch of 96 samples within 48 h (Buyer and Sasser 2012). This high throughput technique implicates the Bligh–Dyer extraction of overnight dried samples and subsequent drying and dissolution of samples in chloroform followed by extraction using a 96 well solid phase extraction column. Elution of phospholipids is performed using 5:5:1 methanol: chloroform: H_2O in a 96 well format glass vial microplate after which drying,

transesterification, and GC analysis are performed subsequently (Buyer and Sasser 2012). For the elution of neutral lipids, chloroform fraction obtained from the SPE column is used (Sharma and Buyer 2015). This method is applicable for both soil and roots (Buyer and Sasser 2012; Sharma and Buyer 2015). The biochemical method analyzing signature fatty acids provides an edge over the error-prone methods such as microscopic visualization of AMF structures. Nevertheless, the incidence of PLFA 16:1ω5cis in bacteria (Nichols et al. 1986) necessitates the need for confirmation of results using microscopic and molecular methods as well.

13.3.6.3 Glomalin

Glomalin is a thermostable glycoprotein formed on the hyphal walls of arbuscular mycorrhizal fungi (Wright and Upadhyaya 1996; Driver et al. 2005). Large quantity of glomalin remains attached to the hyphae and spores and as small as 20% becomes a part of the released fraction (Driver et al. 2005). Upon its release into the soil, it becomes a component of the stable organic matter (Wright and Upadhyaya 1996). Apparently, glomalin exists in two pools. Easily extractable glomalin is believed to be newly formed fraction belonging to young hyphae (Wright and Upadhyaya 1996, 1998; Wright 2000) whereas total glomalin fraction is considered to be a relatively recalcitrant fraction and is often referred to as older glomalin (Lovelock et al. 2004). As it is difficult to extract glomalin from the soil in pure form, Rillig (2004) recommended a new terminology for it, where it was called "glomalin-related soil protein" or "GRSP."

13.3.6.4 Prominence of Glomalin

It plays a key role in soil carbon sequestration as a constituent of the soil organic carbon pool (Rillig et al. 2001) and indirectly by enhancing soil aggregation by acting as a soil particle binding agent (Rillig et al. 2002; Wilson et al. 2009). It has been used as a proficient indicator to elucidate the effect of land use management (Rillig et al. 2003); soil quality and agricultural management approaches (Fokom et al. 2012); assessment of variations in AMF biomass (Krivtsov et al. 2004).

13.3.6.5 Extraction from Soil

Easily extractable glomalin fraction is extracted with 20 mM sodium citrate and 30–60 min autoclaving followed by centrifugation at 5000 xg, and total glomalin fraction is extracted with 50 mM sodium citrate and 60–90 min autoclaving followed by centrifugation at 5000 xg (Wright and Upadhyaya 1996, 1998). Bradford protein assay (Bradford 1976) is extensively used for the quantification of glomalin. The immunoreactive fraction of glomalin is quantified using ELISA (Wright and Upadhyaya 1996).The current extraction protocol rests on the fact that the harsh conditions of temperature and pressure employed for glomalin extraction destroy the vast majority of protein except for glomalin and to get higher recovery depending on soil types, samples may require many cycles of extraction (Agnihotri et al. 2015). The persistence of polyphenols (Whiffen et al. 2007), added glycoproteins and proteins from plant sources (Rosier et al. 2006) in glomalin extracts and their successive binding to Bradford reagent Coomassie brilliant blue G-250 (CBB)

during quantification questions the presently used procedures of its extraction and quantification (Koide and Peoples 2013). Intraradically produced glomalin has been efficaciously used as a signal of AMF root colonization (Rosier et al. 2008).

13.3.7 Earth Worm

Earthworms belong to macrofauna (4–200 mm in size) but some species can reach the dimension attributed to megafauna (>200 mm) (Bachelier, 1986) and are considered soil engineers, as they are able to modify soil structure and features by their etho-physiological action (Gavinelli et al. 2018). Earthworm sampling should preferably be carried out during cool and wet seasons; sampling of dry soils (dry seasons) or of frozen soils should always be avoided. In temperate areas, sampling studies in autumn, spring, and some of the winter months give the best results (Paoletti 1999). Earthworm sampling can be done by hand sorting. It is the traditional method, in which active collection of earthworms from standard soil volumes advocated (Valckx et al. 2011). In detail, this technique consists of extracting a soil bulk (30 × 30 × 20 cm) with a spade fork (Paoletti 1999; Fusaro et al. 2018). Afterwards, a visual examination of soil bulk takes place for 15 min upon a white cloth and each earthworm is picked up. In order to collect deep burrower species, an effective recommendation is the use of an irritant suspension (Bouché 1972; Lee 1985) poured into the soil. The mustard powder water suspension (30 g L^{-1}) acts as an expellant for earthworms and it is a natural substance without toxic or dangerous consequences for the operator and the environment (Pelosi et al. 2009; Valckx et al. 2011). In the humid tropical forests some species are arboriculous and live in suspended soils, such as the soil that accumulates in the leaves rosette of bromeliads, in the tree canopy. These earthworms can be collected by photo-eclectors, a special trap that catches all moving invertebrates on the surface of trunks (Adis and Righi 1989).

13.4 Applications of Soil Health Indicators

Soil health encompasses the physical, chemical, and biological features, but the use of biological indicators is the least well advanced (Griffiths et al. 2018). Hence, for sustainable crop production, the application of different soil health indicators and their analytical techniques used have paramount significance. Lists of application of these indicators along with their analytical methods used in different laboratories are enlisted in Table 13.3.

Table 13.3 Different soil health indicators, analytical techniques and their applications

Soil health indicators	Protocol	References	Application	References
Soil physical health indicators				
Bulk density (Mg m^{-3})	Core sampler	Black (1965)	Soil management, provides information regarding soil compaction that might help in planning of modern farming techniques, Geochemical studies	Sharma and Bhattacharya (2017)
Water holding capacity	Keen Raczkowski cups	Piper (1966)	Availability of water to crops and helps to decide how much quantity of irrigation water to apply for crop production	Bhavya et al. (2018), Water Conservation Factsheet (2015)
Aggregate stability	Wet sieving techniques	Haynes (1993)		
Soil chemical health indicator				
pH	pH meter (supernatant suspension of 1:2 soil to water ratio)	Jackson (1973)	Indication of the acidity or alkalinity of soil and application of soil amendments such as lime/gypsum, crop suitability for location, rough indicator of plant nutrients availability	Kadam (2016), Hanlon (2015)
EC	EC meter (supernatant suspension of 1:2 soil to water ratio)	Jackson (1973)	Helps to know the nature of soil and soluble salts status	Kadam (2016)
CEC	Ammonium acetate method, barium chloride-triethanolamine method	Schollenberger and Dreibelbis (1930), Mehlich (1938)	Indicates the availability of cations in the soil for crop growth and development	Agronomy Fact Sheet Series (2007)
SOC	Walkley–Black or dry combustion/rapid titration method	Walkley and Black (1934)	Main indicator for soil fertility, maintains sustainable productivity and agro ecosystem	Hijbeek (2017), Moharana et al. (2017)
Available N	Micro-Kjeldahl procedure	Subbiah and Asija (1956), Chapman and Pratt (1961)	Imparts greenness to plant, improves growth and yield, and occupies a	Leghari et al. (2016)

			conspicuous place in plant metabolism system	
Available P	Olsen's reagent, Brays solution, ascorbic acid method	Olsen et al. (1954), Bray and Kurtz (1945), Watanabe and Olsen (1965)	Plays important role in plant physiological and biochemical processes and improves plant root architecture	Johnston and Steen (2000), Syers et al. (2008)
Available K	Flame photometrically/ ammonium acetate method	Piper (1966), Hanway and Heidal (1952)	Increases the tolerance level in the plants to biotic and abiotic stresses	Hasanuzzaman et al. (2018)
Available S	Calcium chloride method	Chesnin and Yien (1950)	Plays significant role in biosynthesis oil and enhances oil content in seed crops	Skwierawska et al. (2016)
Micronutrients (Zn, Fe, Mn, and Cu)	DTPA extraction (atomic absorption spectrophotometer)	Lindsay and Norvell (2010)	Acts as a catalyst in various oxidation–reduction reactions, plays integral part in chlorophyll synthesis and activators of several enzymes	Lohry (2007)
Soil biological and biochemical health indicators				
Soil microbial biomass carbon (μg g $soil^{-1}$)	Chloroform fumigation extraction (CFE), dichromate digestion	Nunan et al. (1998), Vance et al. (1987)	Ecosystem services such as carbon cycling, nutrient turnover, production of trace gases, or pollutant degradation	Ananyeva et al. (2008), Makova et al. (2011)
Soil microbial N	Ninhydrin reactive N	Joergensen and Brookes (1990)	----------do----------	----------do-------
Soil respiration (mg CO_2 kg^{-1} soil day^{-1})	Carbon dioxide release	Anderson and Domsch (1980)	----------do----------	
Substrate-induced respiration	Substrate addition (glucose)	Anderson and Domsch (1978)	----------do----------	----------do-------
Enzymes				
Acid and alkaline phosphates	p-Nitrophenol release	Tabatabai (1994), Tabatabai and Bremner (1969)	Organic phosphorus cycling	Cardoso et al. (2013)

(continued)

Table 13.3 (continued)

Soil health indicators	Protocol	References	Application	References
Arylsulfatase	p-Nitrophenol release	Sarathchandra and Perrott (1981)	Organic sulfur cycling	Cardoso et al. (2013)
ß-Glucosidase	p-Nitrophenol release	Tabatabai (1982)	C oxidation	Cardoso et al. (2013)
Dehydrogenase (μgTPF g $soil^{-1}$ day^{-1})	TTC reduction method	Tabatabai (1994), Klein et al. (1971)	Electron transferences in the respiratory chain in living	Cardoso et al. (2013)
FDAse (μg fluorescein g $soil^{-1}$ h^{-1})	Fluorescein diacetate method	Green et al. (2006), Schnürer and Rosswall (1982)	Total microbial activity	Cardoso et al. (2013)
Urease	Non-buffer method	Zantua and Bremner (1975)	Organic N mineralization to ammonia/ ammonium	Cardoso et al. (2013)
Soil organisms				
Mycorrhiza	Microscopic methods (spore density, root colonization)	Gerdemann and Nicolson (1963), Philips and Hayman	Crop production, impact of farming practices, reclamation of stressed ecosystems, drought tolerance	Sharma et al. (2012), Ruiz-Lozano (2003), Berruti et al. (2016), Kabir (2005)
Lipid profiling	Biochemical (16:1ω5cis) PLFA and NLFA PLFA	Sharma and Buyer (2015), Buyer and Sasser (2012), Olsson (1999)	Crop production systems, AMF quality inocula, soil microbial community diversity	Ferrari et al. (2018), Butler et al. (2012)
Earthworm	Hand sorting	Valckx et al. (2011)	Ecological engineers and soil fertility indicators	Van Groenigen et al. (2014)

13.5 Strategies for Management of Health Indicators

The different strategies employed to manage soil health indicators are varied with location, climate, soil type, and land use. But several general principles that focus on sustainable soil health management practices may suit in most of the situations to bring significant improvement in soil health indicators which are increased organic matter, decreased erosion, better water infiltration, more water holding capacity, less subsoil compaction, and less leaching of agrochemicals to groundwater (Rosa and Sobral 2008). The detailed management strategies are listed in Table 13.4.

13.6 Effects of Crop and Soil Management Practices on Soil Health Indicators: Previous Reports

The key crop and soil management practices such as crop rotation, nutrient management, and tillage practices influence the soil physical, chemical, and biological health indicators (Sharma et al. 2010). Crop rotation is a very ancient cultural practice (Howard 1996) that has a strong influence on soil structure, organic matter, and microbial communities (Janvier et al. 2007). Traditionally, it has been used to disrupt disease cycles (Curl 1963) and fix atmospheric nitrogen by legumes for subsequent non-leguminous crops (Pierce and Rice 1998). Sharma et al. (2012) showed the importance of including maize in rotation with soybean under conventional reduced tillage that helped in enhancing soybean yield, AM inoculum load, and organic carbon. Studies on tillage indicate that many critical soil quality indicators and functions can be improved by decreasing tillage intensity (Govaerts et al. 2007a). Compared to conventional tillage, reduced tillage practices offer not only long-term benefits to soil stability, reducing erosion, but also enhance soil microbial diversity (Welbaum et al. 2004; Govaerts et al. 2008). No till practices combined with crop residue retention increase soil organic matter content in the surface layer, improve soil aggregation, and preserve the soil resources better than conventional till practices (Govaerts et al. 2007b). Increased soil organic matter content associated with no till practices not only improves soil structure and water retention but also serves as a nutrient reservoir for plant growth and a substrate for soil microorganisms. Sharma et al. (2012) evaluated the impact of tillage practices and crop sequences on AM fungal propagules and soil enzyme activities in a 10-year long-term field trial in vertisols of soybean–wheat–maize (S–W–M) cropping system where S–M–W or S–W–M–W rotations under reduced-reduced tillage system showed higher soil dehydrogenase activity and fluorescein diacetate hydrolytic activity compared to other combinations. The inclusion of maize in the rotation irrespective of tillage systems showed comparatively higher mycorrhizal and higher phosphatase activities and organic carbon and maintained higher soybean yield. Organic amendments cover a wide range of inputs, including animal manure, solid waste, and various composts, and often improve soil health indicators and productivity. Girvan et al. (2004) and Melero et al. (2006) showed that these amendments, as well as crop residues, resulted in significant increases in total organic carbon

Table 13.4 Strategies of soil health management as per NRCS-USDA (2016)

Management strategies	What does it do?	How does it do?
(I) Conservation crop rotation		
Growing a diverse number of crops in a planned sequence in order to increase soil organic matter and biodiversity in the soil	– Increases nutrient cycling – Manages plant pests (weeds, insects, and diseases) – Reduces sheet, rill, and wind erosion and holds soil moisture – Adds diversity so soil microbes can thrive	– Improves nutrient use efficiency – Decreases use of pesticides – Improves water quality – Conserves water improves plant production
(II) Cover crop		
An un-harvested crop grown as part of planned rotation to provide conservation benefits to the soil	– Increases soil organic matter – Prevents soil erosion and conserves soil moisture – Increases nutrient cycling – Provides nitrogen for plant use, suppresses weeds, and reduces compaction	– Improves water quality and crop production – Conserves water and improves nutrient use efficiency – Decreases use of pesticides – Improves water efficiency
(III) No till		
A way of growing crops without disturbing the soil through tillage	– Increases organic matter and improves water holding capacity of soils – Reduces soil erosion and energy use – Decreases soil compaction	– Conserves water and improves water quality and efficiency – Improves air quality and crop production – Saves renewable resources – Increases productivity
(IV) Mulch tillage		
Using tillage methods where the soil surface is disturbed but maintains a high level of crop residue on the surface	– Reduces soil erosion from wind and rain – Increases soil organic matter, moisture and reduces energy use	– Improves water quality – Conserves water – Saves renewable resources – Improves air quality and crop production
(V) Mulching		
Applying plant residues or other suitable materials to the soil surface to compensate for loss of residue due to excessive tillage	– Reduces erosion from wind and rain and moderates soil temperatures – Increases soil organic matter and conserve soil moisture	– Conserves water, improves air and water quality – Improves crop productivity – Increases crop production

(continued)

Table 13.4 (continued)

Management strategies	What does it do?	How does it do?
	– Reduces dust and control weeds	– Reduces pesticide usage
(VI) Nutrient management		
Managing soil nutrients to meet crop needs while minimizing the impact on the environment and the soil	– Increases plant nutrient uptake – Improves physical, chemical, and biological properties of soil – Budgets, supplies, and conserves nutrients for plant production	– Improves water quality – Improves plant production – Improves air quality

(TOC), Kjeldahl-N, available-P, soil respiration, microbial biomass, and enzyme activities (e.g., protease, urease, and alkaline phosphatase). Microbial diversity and crop yields also increased as compared to conventional management. Khan et al. (2017) reported that integrated nutrient management practices (NPK+FYM) significantly increased soil organic matter and available water holding capacity but decreased the soil bulk density, creating a good soil condition for enhanced crop growth. Microbial population (bacteria, fungi, and actinomycetes) were very responsive to organic manure application. The long-term application of organic manures in rice-brown sarson cropping system increased the index value because it increased the nutrient index (NPKS and micronutrients), microbial index, and crop index of soils. Chemical indicators (pH, EC, and CEC) also improved with integrated nutrient management practices. The use of only chemical fertilizers in the rice–brown sarson cropping system resulted in poor soil microbial index and crop index. Soil pH decreased significantly over the initial values due to the application of organic manures in combination with chemical fertilizers. The lowering of soil pH toward the neutral range favors the availability of different major and micronutrients, viz. N, P, K, Fe, Cu, Mn, Zn, etc. which helps in optimum growth of plants. The highest organic carbon content (0.88%) found in 4 t ha^{-1} manure+ NPK and Zinc at 0.5 kg ha^{-1} applied plot. Hence, there was a great role of INM in augmenting the soil fertility build-up with respect to both major and micronutrients as well as in maintaining soil health indicators (Sur et al. 2010). Crop residue retention along with application of 50% recommended dose of potassium plus seed inoculation of potassium solubilizing bacterial has brought significant improvement in soil physical, chemical, and biological indicators under zero till maize–wheat cropping system and that intern helped in increasing productivity of maize and wheat crops (Raghavendra et al. 2018).

13.7 Conclusion

Soil health indicators are key elements required for maintaining the soil quality. The soil health indicators are dynamic in nature; some of soil health indicators (biological and chemical) are more prone to change in a shorter period whereas some (physical) may take longer period to change due to its management practices. Developing sustainable soil health indicators management practices by using a systematic approach that integrates soil physical, chemical, and biological principles into management practices will help in optimizing the sustainable crop production. There is a need for developing critical levels for some of the soil health indicators to which information is limited. Our research experiments should be planned in such a way that must include three aspects such as soil health indicators restoration, improvement, and maintenance. Systematic research is needed to study soil health indicators for diversity of edaphic, climatic, and management conditions. Conservation agricultural practices such as zero tillage, residue recycling, soil cover management, appropriate crop rotations, and integrated nutrient management practices along with addition of organic amendments have shown the proven benefit to improve soil health indicators.

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