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Biosynthesis and metabolic actions of simple phenolic acids in plants

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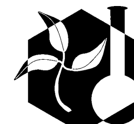

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Abstract The diversity of secondary compounds in the plant kingdom is huge. About 200,000 compounds are known, which are grouped into amines, non-protein amino acids, peptides, alkaloids, glucosinolates, cyanogenic glucosides, organic acids, terpenoids, quinones, polyacetylenes, and phenolics. The group of phenolic compounds consists of polyphenols, oligophenols and monophenols or simple phenolic compounds such as benzoic and cinnamic acids and their hydroxylated derivatives. Among the thousands of compounds present in ecological interactions, simple phenolic acids are the most abundant in soils, and many are described as allelochemicals. Given the physiological and biochemical importance of these compounds, we review their biosynthesis and metabolic actions in plants.

Keywords Allelochemicals · Benzoic acids biosynthesis · Benzoic and cinnamic acids · Lignin · Phenylpropanoid pathway · Secondary metabolites · Shikimate pathway

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Introduction

Didactically, plant metabolism can be divided into two types: primary and secondary. The primary metabolism constitutes processes such as photosynthesis, respiration, the biosynthesis of carbohydrates, lipids and proteins, solute transport, nutrient assimilation and tissue differentiation, which are required for plant growth, reproduction and survival. Primary metabolites are chemical compounds derived from these processes and include amino acids, nucleotides, carbohydrates, fatty acids, and others. The primary metabolism is universal since it is found throughout the plant kingdom. Secondary metabolism constitutes processes such as the biosynthesis of phenylpropanoids, anthocyanins, alkaloids, coumarins, terpenes, tannins, glucosinolates, flavonoids and isoflavonoids, lignans and lignins, among others (Kutchan et al. 2015). Secondary metabolism varies between plant species because the profile of specific secondary metabolites varies between plant species (Shitan 2016). However, secondary compounds have attracted the attention of scientists due to the relevant chemical-biological importance. The chemistry of natural products is a clear example of the application of secondary compounds such as flavorings, perfumes, insecticides, herbicides, antibiotics, polymers and dyes (Hussain et al. 2012). On the other hand, secondary compounds exert relevant ecological functions as protectors against the attack of herbivores and

microorganisms, attractors of pollinating species, in the dispersion of seeds and, especially, in plant-plant interactions (Kennedy and Wightman 2011).

The distribution of secondary compounds in the plant kingdom is huge. About 450,000 secondary compounds are estimated and 200,000 are known and grouped into amines, non-protein amino acids, peptides, alkaloids, glucosinolates, cyanogenic glucosides, organic acids, terpenoids, quinones, polyacetylenes, and phenolics (Wink 2015, 2018). In general, the group of phenolic compounds consists of (1) polyphenols such as lignin and tannins; (2) oligophenols such as flavonoids, stilbenes and coumarins and (3) monophenols (simple phenolic acids) such as benzoic acid derivatives (hydroxybenzoic acids) and cinnamic acid derivatives (hydroxycinnamic acids) (Fig. 1).

Lignin is a complex heteropolymer of simple phenolic acids that confers high mechanical resistance to the plant cell wall (Boerjan et al. 2003). Present in all tracheophytes, lignin allows the transport of water and nutrients from the roots to the leaves by capillarity (Alber and Ehlting 2012; dos Santos et al. 2014). After cellulose, lignin is the most abundant biomolecule on the planet. Lignocellulose had, and still has, a relevant role in the history of mankind, serving as a source of energy and raw material for the construction of houses, tools, weapons and ships (dos Santos et al. 2014). Tannins are polymers of oligophenols whose outstanding feature is their ability to adsorb proteins strongly. The presence of tannins in the vacuoles and waxes inhibits the digestive enzymatic arsenal of pathogens and herbivores (War et al. 2012). In addition to this important biological role, humans

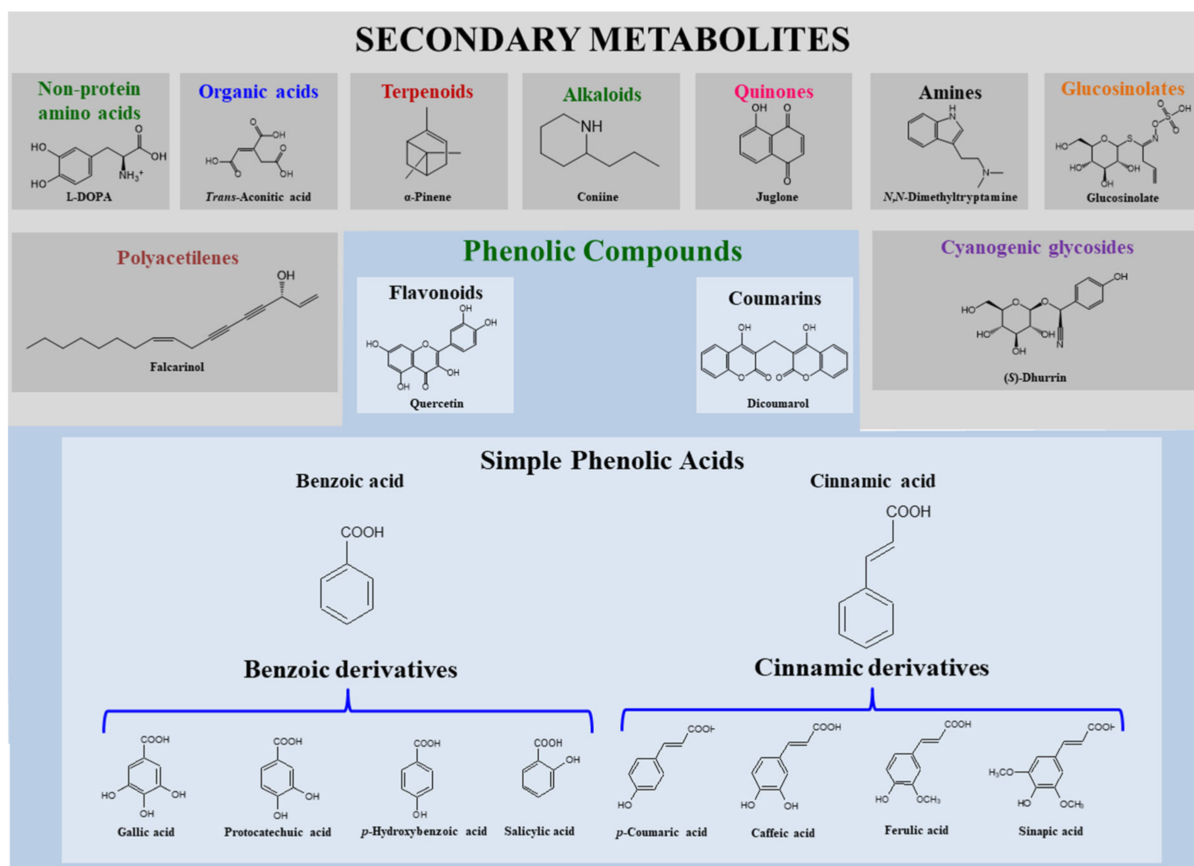


Fig. 1 Distribution of secondary metabolites in the plant kingdom, in which each class is represented by a well-known compound. In evidence are the phenolic compounds, which include flavonoids, coumarins and simple phenolic acids. The latter are the simplest phenolic compounds and are biosynthesized from the benzoic and cinnamic acids

have learned to appreciate their presence in nuts, wines and aged distillates.

Oligophenols are primarily related to the defense mechanisms, and they have been studied for their nutritional and pharmacological properties. Some oligophenols are toxic (for example, furanocoumarins), and cause severe burns when in contact with the skin. However, others are used by humans. For example, cannabinoids, which are psychoactive compounds present in plants of the genus *Cannabis*. Cultivated more than five thousand years ago in the Orient, these plants have been used in religious rituals, medicinal treatments and, more recently, are used as narcotics, as well as for pharmacological and medical applications (Lopez-Vergara et al. 2019; Premoli et al. 2019).

Phenolic compounds have many functions in plants. Some act as defense elements against herbivores and pathogens. Others, such as phenylpropanoids, have antioxidant properties. They protect cell structures against the chain reactions of free radicals that are produced by certain chemical reactions and UV light. Many of them absorb light in the ultraviolet region of the electromagnetic spectrum, acting as protectors against damaging solar radiation. They also play structural roles, interconnecting cell wall polysaccharides and anchoring lignin to the polysaccharide domain, especially in grasses. Due to the economic importance of these plants, hydroxycinnamic acids, especially ferulic acid, have been widely studied, both for their allelopathic and structural properties (which interfere with the digestibility of forage and the production of biofuels from biomass) as well as for their valuable industrial and pharmacological applications (de Paiva et al. 2013; Oliveira et al. 2019).

One of the most relevant and fascinating functions of phenolic compounds is played in the processes of plant communication. Plants communicate in a remarkable way with other plants, and with other organisms. A clear example of the involvement of phenolic compounds in communication is that of anthocyanins (Castañeda-Ovando et al. 2009). Far from being defense compounds, these oligophenols are molecules with great capacity for the absorption of light in different bands of the visible spectrum. Thus, plants use these compounds to define the colors of flowers and fruits. The presence of colors is a strong attraction for pollinating and disseminating agents. In addition to this, an arsenal of chemical compounds is used by plants in subtle interactions. Included in this arsenal are allelopathic compounds (or allelochemicals) stand out, as they are exuded, leached, exhaled or even released by the degradation of roots, leaves, and stems (Fig. 2). Among the thousands of compounds used in ecological interactions, simple phenolic acids are the most abundant in soils (Li et al. 2010). They stimulate or inhibit the growth of neighboring plants favoring the growth of symbiotic species and inhibiting the growth of competing plants. As well as acting on the plants, these compounds act on microbiota attracting symbiotic bacteria, mycorrhizae and inhibiting pathogens and predators.

To date, most reviews about simple phenolic acids have been devoted to specific topics. Thus, given the importance of these compounds in several aspects of plant physiology and biochemistry, the current review covers (1) the biosynthesis of phenylpropanoids and benzoic acids, and (2) the metabolic actions of simple phenolic acids in plants.

Biosynthesis of phenylpropanoids and benzoic acids

Plants produce numerous secondary compounds that contain at least one phenol group in their structures, i.e., a hydroxyl group attached to an aromatic ring. They are classified as phenolic compounds, which constitute a chemically heterogeneous group. Some are soluble in organic solvents, others in water, whereas large polymers, such as lignin may be insoluble (Naczk and Shahidi 2006).

Two fundamental metabolic pathways are involved in the biosynthesis of phenolic compounds in plants: the shikimate pathway and the malonate pathway (Hättenschwiler and Vitousek 2000). Although considered an important source of phenolic compounds in fungi and bacteria, the malonate pathway is less significant in higher plants. Because the shikimate pathway participates in the biosynthesis of most phenolic compounds, the reactions that compose it are highlighted in this review, followed by reactions of the phenylpropanoid pathway and, finally, the biosynthesis of simple benzoic acids.

The shikimate pathway

The shikimate pathway converts carbohydrate precursors, derived from glycolysis and the phosphate

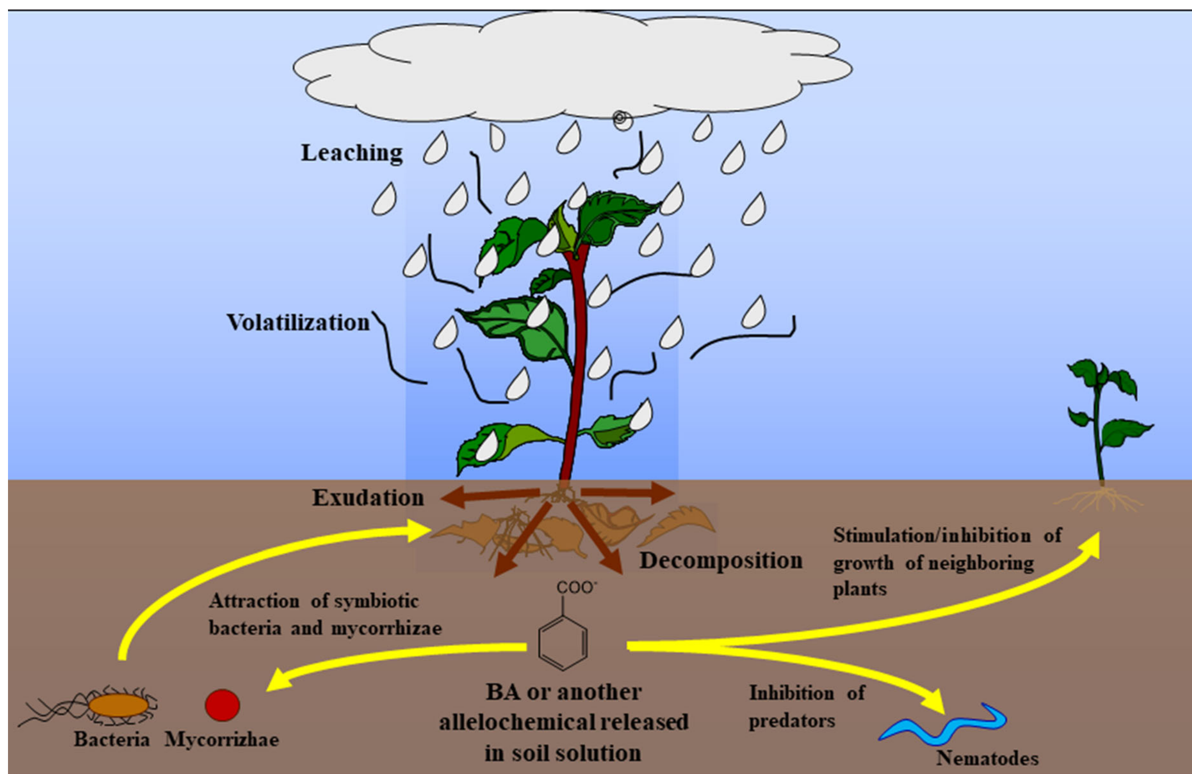


Fig. 2 Plants can release allelochemicals into the environment through exudation, leaching, exhalation or even by the decomposition of roots, stems and leaves. Simple phenolic acids are the most common and important class of allelochemicals present in ecosystems. In soil, these compounds may exert allelopathic effects on neighboring plants or microbiota

pentose pathway, in chorismate, the precursor of aromatic amino acids (Zabalza et al. 2017). It is the metabolic pathway for the biosynthesis of aromatic amino acids (phenylalanine, tyrosine and tryptophan) in plants (Herrmann 1995; Tzin and Galili 2010a). In fact, under normal conditions, up to 20% of the carbon fixed by plants flows into the shikimate pathway, with greater carbon flow under plant stress or rapid growth (Tohge et al. 2013). This pathway is present in bacteria, fungi and plants, but is absent in animals. As a result, their enzymes are targets for the development of drugs against human pathogens and herbicides (Coggins et al. 2003; Bhattacharya and Kumar 2012; Maeda and Dudareva 2012; Coracini and de Azevedo 2014). The biosynthesis of aromatic amino acids occurs in the plastids. However, the three aromatic amino acids and some of the pathway intermediates (for example, shikimate) are exported to the cytosol for the biosynthesis of proteins and other compounds derived from the shikimate pathway. Because of this, some isoenzymes of the shikimate pathway are found in the cytosol. In addition to phenolic compounds, the shikimate pathway provides carbon skeletons for many essential compounds such as indole-3-acetic acid (IAA), tetrahydrofolate (vitamin B9), salicylic acid, plant pigments and quinones responsible for energy transduction in the electron transport chains of thylakoids and mitochondria (Tzin and Galili 2010b; Maeda and Dudareva 2012).

The shikimate pathway consists of a sequence of seven enzymatic steps towards chorismate (Fig. 3), the last precursor that is common to aromatic amino acids (Singh and Christendat 2006). In the first step, erythrose-4-phosphate (E4P), from the non-oxidative branch of the pentose phosphate pathway, and the phosphoenolpyruvate (PEP), produced by the glycolytic pathway, are condensed into 3-deoxy-D-arabinoheptulosonate 7-phosphate (DAHP) by DAHP synthase (DAHPS; EC 2.5.1.54). This enzyme requires Mn^{2+} and reduced thioredoxin and, therefore, links the carbon flux in the shikimate pathway to the electron flow in photosystem I (Tzin and Galili 2010a).

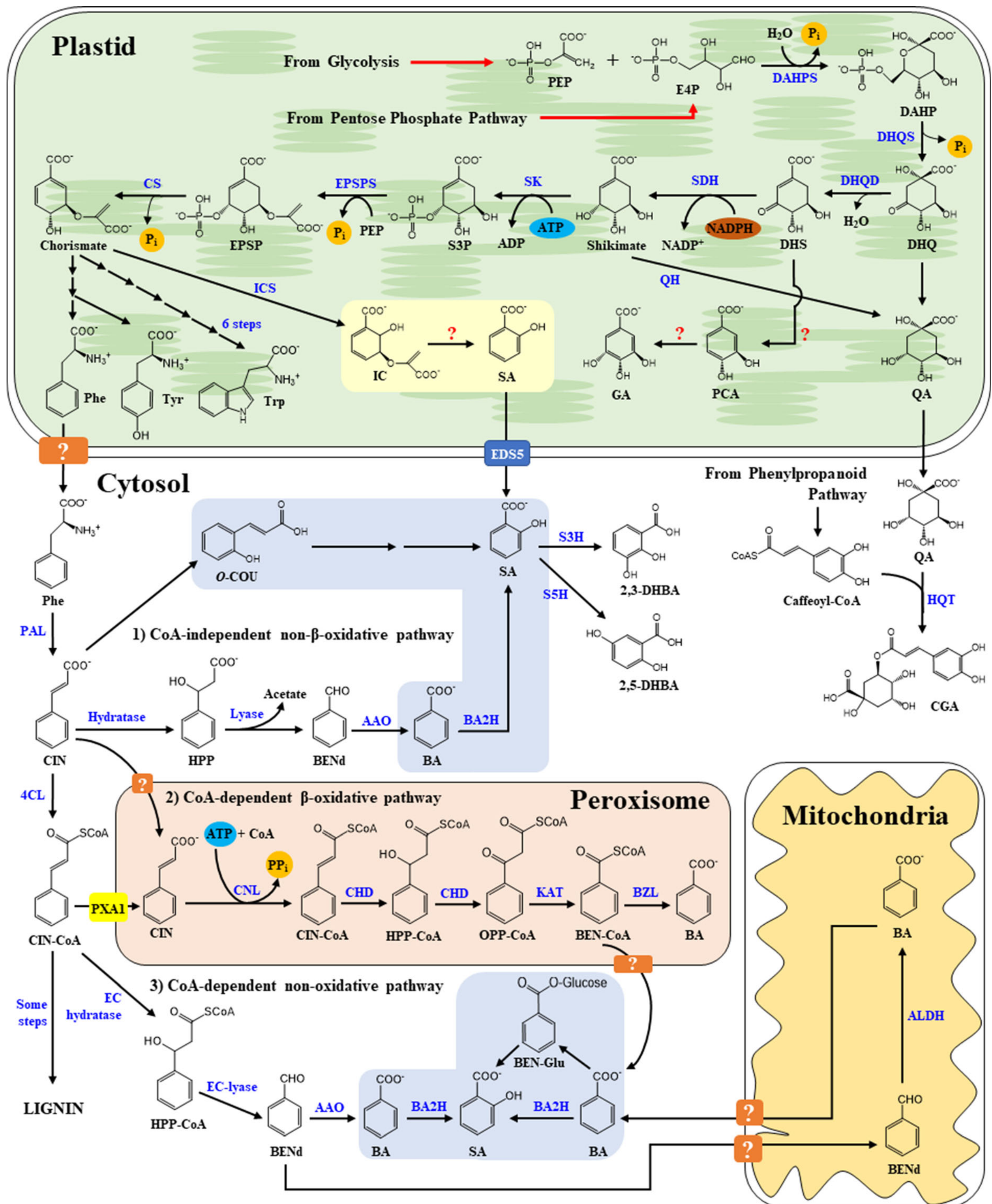
According to their amino acid sequence homology and molecular mass, DAHPs can be classified into class I and class II enzymes. The presence of a domain with chorismate mutase (CM) activity is used to classify the microbe DAHPs in class I (Tohge et al. 2013). The class II DAHPs in plants, in turn, do not have an associated CM activity.

In the second step, DAHP is cyclized, forming 3-dehydroquinate (DHQ) in a reaction catalyzed by DHQ synthase (DHQS; EC 4.2.3.4). The reaction involves the steps of oxidation, phosphate elimination, reduction, ring opening and internal aldol condensation, although there is no formation of by-products. In plants, DHQS is monofunctional, and its structural characterization was first performed for *Actinidia chinensis* (New Zealand kiwifruit); it is a homodimeric protein in which each monomer has a NAD^{+} binding site (Mittelstädt et al. 2013).

In the third step, DHQ undergoes two reactions that are combined and catalyzed by the bifunctional enzyme 3-dehydroquinate dehydratase/shikimate dehydrogenase (DHQD/SDH; EC 4.2.1.10 and EC 1.1.1.25, respectively) to form shikimate (Herrmann and Weaver 1999). DHQD (also known as dehydroquinase) initially catalyzes the dehydration of DHQ to 3-dehydroshikimate (DHS), which is subsequently reduced to shikimate by SDH with the use of NADPH. The crystallographic structure of the enzyme complex of *Arabidopsis thaliana* revealed that the active sites of DHQD and SDH are arranged face-to-face to channel the transfer of the DHS intermediate and reduce its diffusion outwards from the complex (Singh and Christendat 2007). Channeling of the substrate between DHQD and SDH ensures the constant flow of precursors in the biosynthesis of chorismate. In addition, the significantly higher activity of DHQD relative to SDH (about tenfold) makes it possible to efficiently convert DHQ to shikimate (Tzin and Galili 2010a). In many plant species, a single gene encodes DHQD/SDH. The gene contains a coding sequence (Tohge et al. 2013) a signal peptide to target the enzyme for plastids (Bischoff et al. 2001; Ding et al. 2007a, b; Singh and Christendat 2007). However, in the *Nicotiana tabacum* genome, two genes encoding DHQD/SDH were found: one cytosolic and another plastidic (Ding et al. 2007a, b). This dichotomy seems important in regulating the biosynthesis of chlorogenic acid (CGA) and other benzoic acid (BA) derivatives from shikimate. Bifunctional enzymes regulate the biosynthesis of metabolites at branch points of a metabolic pathway. DHQD/SDH is the only bifunctional enzyme in the shikimate pathway. It is located at a point of branching that leads to the biosynthesis of quinate and other benzoates, such as protocatechuic and gallic acids (Ghosh et al. 2012).

In the fifth step, shikimate is activated to shikimate 3-phosphate (S3P) by the action of shikimate kinase (SK; EC 2.7.1.71). This reaction, which requires divalent cations such as Mg^{2+} and Mn^{2+}, transfers a phosphate group from the ATP to the C3 hydroxyl group of shikimate. Belonging to the family of nucleotide monophosphate (NMP) kinases, SK consists of core, lid, shikimate-, and nucleotide-binding domains (Maeda and Dudareva 2012). The number of isoforms of the enzyme is species-dependent; *Solanum lycopersicum, A. thaliana* and *Oryza sativa* have one, two and three genes encoding SK, respectively. It has been suggested that SK can regulate flow through the shikimate pathway because its activity is influenced by cellular energy status (Tohge et al. 2013). An earlier in vitro study performed with SK purified from *Spinacia oleracea* showed that the enzyme is only slightly inhibited by ADP, but strongly inhibited after adjustment of the energy charge with the aid of a myokinase (Schmidt et al. 1990).

The sixth step, catalyzed by 5-enolpyruvylshikimate 3-phosphate (EPSP) synthase (EPSPS; EC 2.5.1.19), is the condensation of S3P with a second molecule of PEP to produce EPSP and inorganic phosphate. This is an unusual reaction because, unlike most enzymes that use PEP, the transfer of an enolpyruvyl moiety from PEP to S3P occurs through the breakdown of the C–O bond, and not of the P–O bond (Schönbrunn et al. 2001). The EPSPS, also known as 3-phosphoshikimate 1-carboxyvinyltransferase, consists of two domains, each containing three copies of a βαβαββ-folding unit (Stallings et al. 1991). The binding of shikimate, the first substrate, induces a conformational change that creates an active site in the inter-domain of EPSPS. The EPSPS is inhibited by a structural analogue of PEP, the glyphosate [(*N*-phosphonomethyl) glycine], a broad-spectrum herbicide widely used to control weeds. Due to the structural similarity with PEP, glyphosate binds to the enzyme-S3P complex at the same PEP binding site, being a competitive inhibitor *versus* PEP and uncompetitive inhibitor *versus* S3P (Schönbrunn et al. 2001). Sensitivity to glyphosate has been used as a criterion for the



Plastid
From Glycolysis
From Pentose Phosphate Pathway
PEP
E4P
DAHPS
DAHP
DHQS
DHQD
DHQ
SDH
DHS
NADPH
NADP+
Shikimate
SK
ATP
ADP
S3P
EPSPS
PEP
EPSP
CS
Chorismate
ICS
6 steps
QH
IC
SA
GA
PCA
QA
Phe
Tyr
Trp
EDS5
Cytosol
From Phenylpropanoid Pathway
Caffeoyl-CoA
HQT
CGA
O-COU
S3H
S5H
2,3-DHBA
2,5-DHBA
PAL
1) CoA-independent non-β-oxidative pathway
Hydratase
HPP
Lyase
Acetate
BENd
AAO
BA
BA2H
CIN
4CL
2) CoA-dependent β-oxidative pathway
Peroxisome
CIN-CoA
PXA1
CNL
CHD
KAT
BZL
HPP-CoA
OPP-CoA
BEN-CoA
Mitochondria
ALDH
3) CoA-dependent non-oxidative pathway
Some steps
EC hydratase
EC-lyase
BEN-Glu
LIGNIN

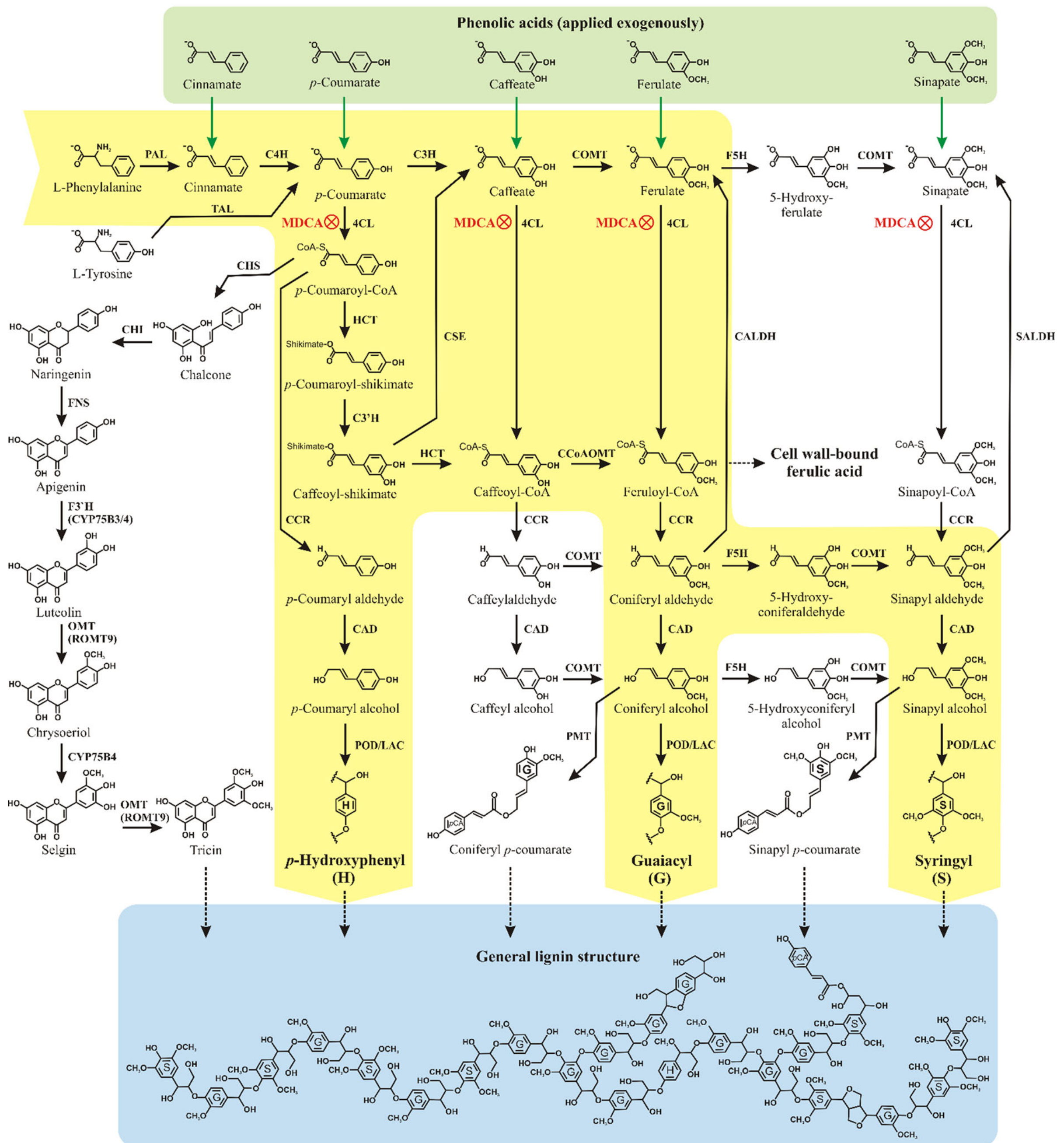
◀**Fig. 3** Biosynthesis of aromatic amino acids (tryptophan, tyrosine and phenylalanine) and benzoic acids in plants. The shikimate pathway is the metabolic pathway for the biosynthesis of aromatic amino acids and occurs in plastids. The biosynthesis of benzoic acids involves four cell compartments: plastid, cytosol, mitochondria and peroxisome. The biosynthesis of quinic (QA), protocatechuic (PCA) and gallic (GA) acids occurs in the plastid directly from the intermediates of the shikimic acid pathway. The carrier utilized to export phenylalanine from the plastid to the cytosol is not yet known. Phenylalanine (Phe) is utilized to biosynthesis of benzoic acids from the phenylpropanoid pathway. CoA-dependent β-oxidative pathway generates benzoic acid (BA) in the peroxisomes. Non-β-oxidative pathways are cytosolic. The transport of benzoyl-CoA (BEN-CoA), or its free form, from the peroxisomes to the cytosol is also unknown. Mitochondrial aldehyde dehydrogenases (ALDH) convert benzaldehydes (BENd) to acids forms in CoA-independent and CoA-dependent pathways. Salicylic acid (SA) can be biosynthesized from isochorismate (IC) produced in the shikimate pathway or from cinnamic acid (CIN) produced from the phenylpropanoid pathway. The route that uses IC as a precursor is plastidial (yellow background). After their biosynthesis in plastids, SA can be exported to the cytosol via the enhanced disease susceptibility 5 (EDS5) carrier. The route that uses CIN as a precursor is cytosolic and involves the reactions of CoA-dependent β-oxidative, CoA-independent non-oxidative, and CoA-dependent non-oxidative pathways. In some cases, BA acts as an intermediate (blue background). Phosphoenolpyruvate (PEP); erythrose 4-phosphate (E4P); 3-deoxy-D-arabino-heptulosonate 7-phosphate (DAHP); DAHP synthase (DAHPS); 3-dehydroquinate (DHQ); 3-dehydroquinate synthase (DHQS); 3-dehydroshikimate (DHS); 3-dehydroquinate dehydratase/shikimate dehydrogenase (DHQD/SDH); shikimate kinase (SK); shikimate 3-phosphate (S3P); 5-enolpyruvylshikimate 3-phosphate (EPSP); EPSP synthase (EPSPS); chorismate synthase (CS); tyrosine (Tyr); tryptophan (Trp); IC synthase (ICS); chlorogenic acid (CGA); hydroxycinnamoyl-CoA quinate: hydroxycinnamoyl transferase (HQT); 2,3-dihydroxybenzoic acid (2,3-DHBA); 2,5-dihydroxybenzoic acid (2,5-DHBA); SA 3-hydroxylase (S3H); SA 5-hydroxylase (S5H); *o*-coumaric acid (*O*-COU); cinnamoyl-CoA ligase (CNL); cinnamoyl-CoA (CIN-CoA); phenylalanine ammonia-lyase (PAL); 4-coumarate-CoA: ligase (4CL); 3-hydroxy-3-phenylpropionic acid (HPP); CIN-CoA hydratase/CIN-CoA dehydratase (CHD; 3-hydroxy-3-phenylpropionyl-CoA (HPP-CoA); 3-oxo-3-phenylpropanoyl-CoA (OPP-CoA); 3-ketoacyl-CoA thiolase (KAT); benzoyl-CoA ligase (BZL); enoyl-CoA hydratase (EC-hydratase); enoyl-CoA (EC-lyase); aldehyde oxidase (AAO); benzoic acid 2-hydroxylase (BA2H); benzoyl-glucose (BEN-Glu); aldehyde dehydrogenase (ALDH)

classification of EPSPS from different organisms. Class I enzymes are present in all plants and most of the bacteria, organisms that have a glyphosate sensitive EPSPS. In turn, some bacteria such as *Agrobacterium* sp. strain CP4 have a glyphosate-insensitive class II EPSPS (Maeda and Dudareva 2012).

Finally, chorismate synthase (CS; EC 4.2.3.5) dephosphorylates EPSP to produce chorismate. Biochemically unique in nature, the CS reaction occurs via *trans*-1,4-elimination of the 3-phosphate and C6-pro-R hydrogen from the EPSP (Kitzing et al. 2004). Although there is no general change in the redox state during the reaction, the enzyme has an absolute requirement for reduced flavin nucleotide ($FMNH_2$), which is not consumed in the reaction. The initial transfer of electrons from $FMNH_2$ to the substrate facilitates the cleavage of the C–O bond (release of phosphate) and the abstraction of hydrogen (Macheroux et al. 1999). In fungi, the CS is a bifunctional enzyme because it presents an NADPH-dependent flavin reductase activity that provides the $FMNH_2$ which is necessary for the reaction. The enzyme of bacteria and plants does not possess the ability to use NADPH to reduce oxidized FMN, which in turn must be provided by photon reduction by blue light or activity of a flavin reductase non-associated to the enzyme.

The phenylpropanoid pathway

As described previously, the shikimic acid pathway generates the aromatic amino acids tryptophan, tyrosine and phenylalanine (Fig. 3). Alkaloids, glucosinolates, phytoalexins and auxins are biosynthesized from tryptophan, while tocopherols, cyanogenic glycosides, suberin and hydroxycinnamic acids, among others, are formed from tyrosine. One of the most important derivatives of tyrosine is plastoquinone, which is required for photosystem II activity and is a cofactor for phytoene desaturase. Most of the simple phenolic compounds biosynthesized in plants come from the aromatic amino acid phenylalanine. Phenylpropanoids are a diverse group of compounds derived from phenylalanine and tyrosine after deamination (Fig. 4). From phenylalanine and tyrosine, lignin biosynthesis involves successive hydroxylations and methylations of the aromatic ring and side-chain modifications to yield lignin monomers (Boerjan et al. 2003; Vanholme et al. 2010). Despite commonality in pathway structure, different plant species show different regulatory features and distinct spatial and topological characteristics; recent advances in mathematical modelling have improved the comprehension of the phenylpropanoid pathway (Cesarino et al. 2016; Faraji et al. 2018).



The phenylpropanoid pathway begins with the reaction of phenylalanine ammonia-lyase (PAL; EC 4.3.1.5) which catalyzes the deamination of phenylalanine to form *trans*-cinnamic acid (Fig. 4). The PAL reaction is strategically located at a point of branching between primary and secondary metabolism. Thus, the reaction is an important regulatory step in the formation of many phenolic compounds. In the next step, *t*-cinnamic acid is hydroxylated at the C4 position of the aromatic ring by the action of cinnamate 4-hydroxylase (C4H, EC 1.14.13.11), forming *p*-coumaric acid. Alternatively, in grasses, *p*-coumaric acid can also be formed from tyrosine by tyrosine ammonia-lyase (TAL), bypassing the

◀**Fig. 4** Phenylpropanoid pathway and mode of action of hydroxycinnamic acids in lignification. Phenylalanine ammonia-lyase (PAL); tyrosine ammonia-lyase (TAL); cinnamate 4-hydroxylase (C4H); 4-coumarate-CoA: ligase (4CL); 3,4-(methylenedioxy)cinnamic acid (MDCA); *p*-hydroxycinnamoyl-CoA: quinate/shikimate *p*-hydroxycinnamoyl transferase (HCT); *p*-coumaroyl-shikimate/quinate 3-hydroxylase (C3′H); 4-coumarate 3-hydroxylase (C3H); caffeoyl shikimate esterase (CSE); caffeoyl-CoA 3-*O*-methyl transferase (CCoAOMT); cinnamoyl-CoA reductase (CCR); coniferyl aldehyde dehydrogenase (CALDH); sinapyl aldehyde dehydrogenase (SALDH); ferulate (coniferil aldehyde/alcohol) 5-hydroxylase (F5H); cinnamyl (caffeate) *O*-methyltransferase (COMT); cinnamyl alcohol dehydrogenase (CAD); *p*-coumaroyl-CoA:monolignol transferase (PMT); peroxidase (POD); laccase (LAC); syringyl (S); guaiacyl (G); *p*-hydroxyphenyl (H); chalcone synthase (CHS); chalcone isomerase (CHI); flavone synthase II (FNS); flavonoid 3′-monooxigenase (F3′H); 3′-*O*-methyltransferase (OMT, ROMT9); chrysoeriol 5′-hydroxylase (CYP75B4); 3′,5′-*O*-methyltransferase (OMT, ROMT9)

reaction of C4H (Barros et al. 2016). C4H is a cytochrome P450-dependent monooxygenase and composes a multi-enzyme complex with the enzyme *p*-coumaroyl (shikimate/quinate) 3-hydroxylase (C3′H; EC 1.14.13.36), which hydroxylates the *p*-coumaroyl residue or free *p*-coumaric acid at C3 to produce the residue caffeoyl or caffeic acid, respectively. It has recently been reported that a bifunctional cytosolic ascorbate peroxidase catalyzes the direct 3-hydroxylation of *p*-coumaric acid to caffeic acid (C3H; Fig. 4) (Barros et al. 2019). This enzyme connects the lignin biosynthesis to stress responses, and it appears to be associated with the detoxification of stress-induced reactive oxygen species (ROS), plant defense, and acclimation pathways.

For monolignol biosynthesis, *p*-hydroxycinnamate-CoA ligase (4CL, EC 6.2.1.12) catalyzes the ATP-dependent activation of *p*-coumaric acid to *p*-coumaroyl-CoA, which is a point of divergence of the pathway and may follow different destinations (Fig. 4). The biosynthesis of monolignols uses *p*-coumaroyl-CoA as an intermediate to produce *p*-coumaryl aldehyde in a reaction catalyzed by cinnamoyl-CoA reductase (CCR; EC 1.2.1.44). *p*-Coumaryl aldehyde is immediately converted to *p*-coumaryl alcohol by the action of cinnamyl alcohol dehydrogenase (CAD, EC 1.1.1.195), the less abundant monolignol in the lignin structure in higher plants. Alternatively, *p*-coumaroyl-CoA follows for the biosynthesis of other phenylpropanoids. In this case, it is transesterified with shikimic acid or quinic acid by the action of the *p*-hydroxycinnamoyl-CoA:quinate/shikimate *p*-hydroxycinnamoyl transferase (HCT; EC 2.3.1.133). The *p*-coumaroyl shikimate is the substrate for *p*-coumarate 3-hydroxylase (C3H), which hydroxylates the residue *p*-coumaroyl of the molecule in C3 to produce caffeoyl-shikimate/quinate. The caffeoyl-shikimate/quinate is transesterified again with CoA, by the action of HCT, forming caffeoyl-CoA, which is methylated on the C3 hydroxyl by the caffeoyl-CoA *O*-methyltransferase (CCoAOMT; EC 2.1.1.104) producing feruloyl-CoA, the activated form of ferulic acid. Next, feruloyl-CoA is reduced by CCR to form coniferyl aldehyde.

In the biosynthesis of monolignols, coniferyl aldehyde follows two distinct pathways: reduction to yield coniferyl alcohol or methoxylation followed by reduction to produce sinapyl alcohol (Fig. 4). The reduction of coniferyl aldehyde is catalyzed by CAD, forming coniferyl alcohol, the most abundant monolignol. Coniferyl aldehyde may also be hydroxylated by the action of coniferyl aldehyde 5-hydroxylase (CAld5H; EC 1.14.13.-) and, then, methoxylated in C5 by COMT to form sinapyl aldehyde. Sinapyl alcohol dehydrogenase (SAD), an enzyme analogous to CAD, reduces sinapyl aldehyde to sinapyl alcohol, the second most abundant monolignol. An analogous sequence, i.e., cleavage followed by reduction of the aldehyde to alcohol, produces the *p*-coumaryl alcohol, another constituent of the lignin polymer. Oxidative polymerization of the *p*-coumaryl, coniferyl and sinapyl alcohols (or monolignols), in reactions catalyzed by peroxidases (POD) and laccases (LAC), respectively, generate *p*-hydroxyphenyl (H), guaiacyl (G) and siringyl (S), monomeric lignin units (Boerjan et al. 2003; Vanholme et al. 2010; Umezawa 2018). Commelinid monocotyledon have lignin polymers partially acylated by *p*-coumarate via the *p*-coumaroyl-CoA:monolignol transferase (PMT; EC 2.3.1) (Withers et al. 2012; Petrik et al. 2014).

In addition to lignin, the phenylpropanoid pathway provides free and simple phenolic acids such as *p*-coumaric, caffeic, ferulic and sinapic acids. Ferulic and sinapic acids are produced by the oxidation of coniferyl aldehyde and sinapyl aldehyde by coniferyl/sinapyl aldehyde dehydrogenase (CALDH/SALDH; EC 1.2.1.68), respectively (Nair et al. 2004; dos Santos et al. 2014). *p*-Coumaric acid is an intermediate of the

pathway, produced from *t*-cinnamic acid by C4H. This same enzymatic complex catalyzes the biosynthesis of caffeic acid formed by the hydroxylation of *p*-coumaric acid (de Oliveira et al. 2017). The caffeoyl shikimate may be cleaved by the action of caffeoyl shikimate esterase (CSE; EC 3.1.1) producing caffeic acid (Fig. 4) (Vanholme et al. 2013). *p*-Coumaroyl-CoA may act as a precursor for the biosynthesis of tricin (3′,5′-dimethoxyflavone), which has been described as the first non-monolignol monomer in monocot lignin (Lan et al. 2015). The complete pathway of tricin biosynthesis (Fig. 4) was recently elucidated in *O. sativa* (Lam et al. 2015). In the first committed step, chalcone synthase condenses three molecules of malonyl-CoA with a molecule of *p*-coumaroyl-CoA to produce 2′,4,4′,6′-tretrahydroxy-chalcone, a chalcone. This enzyme may also act on dihydro-*p*-coumaroyl-CoA forming phloretin (Gosch et al. 2010), a flavonoid with anti-arthritic and anti-inflammatory activities (Wang et al. 2016). Further, chalcone isomerase converts chalcone to naringenin (a flavanone), which is metabolized into apigenin by flavone synthase II (CYP93G1), a cytochrome P450 enzyme. The subsequent hydroxylation of apigenin by flavonoid 3′-monooxygenase (F3′H, CYP75B3/CYP75B4) produces luteonin, which is converted to chrysoeriol by a 3′-*O*-methyltransferase (OMT, e.g., ROMT9). Finally, the hydroxylation of chrysoeriol by chrysoeriol 5′-hydroxylase (CYP75B4) generates selgin that is methylated in tricin by a 3′,5′-OMT (Lam et al. 2015; Poulev et al. 2019). In addition to its structural role as a component of lignin in monocots, tricin has antioxidant, anti-aging, anticancer and cardioprotective properties (Ogo et al. 2013; Lan et al. 2015).

It is noteworthy that phenylpropanoids are basic units for the formation of more complex phenolic compounds such as oligophenols and polyphenols, or also alkaloids, cyanogenic glycosides and glucosinolates (Wink 2010). They can also be oxidized to produce simpler compounds, such as those derived from benzoic acid, by eliminating side chain carbons (Wildermuth 2006).

The biosynthesis of benzoic acids

From a chemical point of view, benzoic acids are organic acids whose carboxyl group is attached to an aromatic ring. From the metabolic point of view, they are extremely versatile compounds that can act as precursors to primary and secondary metabolites. For example, they can produce phytohormones, attractant compounds for pollinators and seed dispersers, defense compounds and electron carriers. In addition, some of them also have pharmacological properties or are used as a raw material for organic synthesis (Effmert et al. 2005; Wildermuth 2006; Qualley et al. 2012; Widhalm and Dudareva 2015).

Several metabolic pathways produce benzoic acids and their derivatives in plants. Some of these have not yet been fully elucidated, although significant progress has been made in recent years. Here, we describe the main metabolic pathways involved in the biosynthesis of benzoic acid derivatives, especially benzoic, *p*-hydroxybenzoic (4-hydroxybenzoic), protocatechuic (3,4-dihydroxybenzoic), gallic (3,4,5-trihydroxybenzoic), salicylic (2-hydroxybenzoic) and chlorogenic acids.

The biosynthesis of benzoic acids can occur from intermediates of the shikimate or phenylpropanoid pathways (Fig. 3). In the first case, 3-dehydroquinate, 3-dehydroshikimate, shikimate and chorismate act as precursors, and the carboxyl group of these intermediates is maintained in the structure of the benzoic acid formed in the reaction. Quinic acid can be produced by the reduction of 3-dehydroquinate or dehydration of shikimate, reactions which are catalyzed by quinate dehydrogenase (QDH) and quinate dehydratase (QD), respectively (Guo et al. 2014). In turn, hydroxycinnamoyl-CoA quinate:hydroxycinnamoyl transferase (HQT) can esterify quinic acid with caffeoyl-CoA from the phenylpropanoid pathway to produce chlorogenic acid, an antioxidant and anti-herbivore (Niggeweg et al. 2004). In addition, quinic acid and shikimate can be esterified to *p*-coumaric acid in the phenylpropanoid pathway. The formation of these transient esters is a key step in lignin biosynthesis (Boerjan et al. 2003).

It is suggested that 3-dehydroshikimate can be converted to gallic acid, although the enzymes involved in this route are unknown. It is possible that 3-dehydroshikimate is directly oxidized or undergoes dehydration followed by hydroxylation to form gallic acid (Muir et al. 2011). In the latter case, the metabolite formed is protocatechuic acid. Chorismate acts as a precursor for biosynthesis of salicylic acid, 2,3-dihydroxybenzoic (2,3-DHBA), 2,5-dihydroxybenzoic (2,5-DHBA) acids and phylloquinone. The

common intermediate in the biosynthesis of these compounds is isochorismate, which is formed by the isomerization of chorismate by isochorismate synthase (ICS). Due to the important biological role, the biosynthesis of salicylic acid and its derivatives is discussed in a specific section of this review.

Benzoic acids can also be produced from the phenylpropanoid pathway intermediates and, in this case, their carboxyl group is formed by shortening the propanoid side chain (Fig. 3). Initially, phenylalanine is transported to the cytosol, where it is deaminated to *t*-cinnamic acid by PAL, the first enzyme in the phenylpropanoid pathway. From *t*-cinnamic acid, three routes have been proposed for the shortening of the propanoid side chain and formation of the carboxyl group: (1) the CoA-independent non-β-oxidative pathway, (2) the CoA-dependent β-oxidative pathway and (3) the CoA-dependent non-oxidative pathway (Fig. 3) (Wildermuth 2006; Widhalm and Dudareva 2015).

The CoA-dependent β-oxidative pathway occurs in peroxisomes and is similar to the β-oxidation of fatty acids and branched-chain amino acids (Wildermuth 2006). In the first step of the pathway, *t*-cinnamic acid is activated by esterification with CoA by cinnamoyl-CoA ligase (CNL). Then, the cinnamoyl-CoA hydratase/cinnamoyl-CoA dehydrogenase (CHD) bifunctional complex catalyzes the production of 3-oxo-3-phenylpropanoyl-CoA from cinnamoyl-CoA (Qualley et al. 2012), with 3-hydroxy-3-phenylpropanoyl-CoA being an intermediate in the process. The last step of the pathway, carried out by 3-ketoacyl thiolase (KAT), is characterized by the elimination of the acetyl-CoA group through the cleavage of the β-keto thioester intermediate with concomitant production of benzoyl-CoA (Widhalm and Dudareva 2015). *p*-Hydroxybenzoic acid is also biosynthesized by the β-oxidative pathway, but the precursor is *p*-coumaroyl-CoA and not *p*-cinnamoyl-CoA. To date, it is not known as substrates and products of this pathway are transported into and out of the peroxisomes. However, it is known that an ABC PXA1 transporter with ATP-stimulated acyl-CoA thioesterase activity is required for the transport of substrates into the peroxisomes in *A. thaliana* (Fig. 3) (de Marcos Lousa et al. 2013). Thus, it is likely that the acyl-CoA substrates are de-esterified before being transported into the peroxisomes. This also requires a CoA ligase to reactivate the substrate for β-oxidation in the lumen of the organelle (de Marcos Lousa et al. 2013). Benzoyl-CoA products are also likely to be de-esterified in the peroxisomes prior to their transport to the cytosol by an unknown transporter, or only passing into the cytosol by simple diffusion through the membrane. In fact, a benzoyl-CoA ligase (BZL) appears to produce benzoic acid from benzoyl-CoA (Dempsey et al. 2011).

The CoA-independent non-oxidative pathway begins with the hydration of the free hydroxycinnamic acids to form 3-hydroxy-3-phenylpropanoic intermediates, which undergo lateral chain degradation via reverse aldol reaction, releasing acetate and benzaldehyde (Fig. 3). In *Vanilla planifolia*, a 4-hydroxybenzaldehyde synthase (4HBD) was purified and shown to convert *p*-coumaric acid to 4-hydroxybenzaldehyde and acetate without the addition of CoA (Podstolski et al. 2002). In addition, Gallage et al. (2014) identified a hydratase/lyase enzyme, also termed vanillin synthase (*Vp*VAN), which converts ferulic acid directly to vanillin. Given its commercial importance, vanillin biosynthesis is described in more detail later.

The CoA-dependent non-oxidative pathway begins with CoA esters and not with free acids, such as in CoA-independent non-oxidative pathway (Fig. 3). The CoA esters are converted to 3-hydroxy-3-phenylpropanoyl-CoA intermediates, which undergo chain shortening via a reverse aldol reaction with the release of acetyl-CoA and benzaldehyde. An enoyl-CoA hydratase/lyase (EC-hydratase/lyase) appears to participate in the production of benzaldehydes from esterified forms (Widhalm and Dudareva 2015). Cytosolic aldehydes oxidases (AAO) and/or mitochondrial aldehyde dehydrogenases (ALDH) convert benzaldehydes to acid forms in the CoA-independent and CoA-dependent pathways.

Biosynthesis of vanillin

Vanillin (4-hydroxy-3-methoxybenzaldehyde) is the most popular flavor compound in the world. The main natural resource of vanillin is the pod of *V. planifolia* (Orchidaceae family), where it accumulates as vanillin glucoside (Gallage et al. 2014; Gallage and Møller 2015; Kundu 2017; Yang et al. 2017). Substantial effort has been made in the last two decades to completely elucidate the metabolic pathway that leads to vanillin biosynthesis in plants (Fig. 5), with some disagreement reported. In *V. planifolia*, the

biosynthesis of vanillin occurs in plastids or in re-differentiated plastids termed "phenyloplasts". More recently, it has been shown that the all enzymes required for the conversion of [^{14}C]-phenylalanine to [^{14}C]-vanillin glucoside are confined within that organelle (Gallage et al. 2018). Thus, it is widely accepted that hydroxycinnamic acids (cinnamic, *p*-coumaric, caffeic and ferulic acids) are precursors of vanillin biosynthesis. Other studies also report the important participation of phenylpropanoid pathway enzymes, as well as a chain shortening process to convert C6–C3 compounds (such as hydroxycinnamic acids) into C6–C1 compounds (such as vanillin).

In one of the possible pathways identified (Fig. 5), vanillin synthase (*Vp*VAN) directly catalyzes the formation of vanillin and vanillin glucoside from ferulic acid and ferulic acid glucoside, respectively (Gallage et al. 2014) (Fig. 5). This reaction occurs in the absence of ATP and NAD$^+$, cofactors that would be required for a chain shortening CoA-dependent β-oxidative pathway. Therefore, in this case, vanillin biosynthesis from ferulic acid occurs through a CoA-independent non-β-oxidative pathway. In another possibility (Fig. 5), ferulic acid is successively converted to feruloyl-CoA, vanillyl-CoA, and finally vanillin (Zenk 1965). However, the enzymes involved in this CoA-dependent β-oxidative pathway have not been identified.

A CoA-dependent non-β-oxidative pathway to vanillin biosynthesis has also been suggested (Fig. 5), in which *p*-coumaric acid is firstly converted to 4-hydroxybenzaldehyde by 4-hydroxybenzaldehyde synthase (4-HBS) (Podstolski et al. 2002). Then, hydroxylation of the 4-hydroxybenzaldehyde aromatic ring at the C3 position leads to the production of 3,4-dihydroxybenzaldehyde (or protocatechuic aldehyde). In the last step, methylation of C3-hydroxyl of 3,4-dihydroxybenzaldehyde, catalyzed by a multifunctional *O*-methyltransferase (OMT), produces vanillin (Pak et al. 2004). This OMT, however, has higher catalytic efficiency with caffeoyl aldehyde and 5-OH-coniferaldehyde as substrates and seems to be primarily involved in the lignin biosynthesis.

Some evidence also supports vanillin biosynthesis through another CoA-dependent non-β-oxidative pathway. In *Hypericum androsaemum*, a bifunctional cinnamoyl-CoA hydratase/lyase catalyzes the hydration of cinnamoyl-CoA to 3-hydroxy-3-phenylpropionyl-CoA followed by retro-aldol cleavage of the reaction product to benzaldehyde and acetyl-CoA (Ahmed et al. 2002). Similarly, a gene encoding an enoyl-CoA hydratase/lyase was identified in *Pseudomonas fluorescens* (strain AN103) (Gasson et al. 1998). This enzyme can catalyze the hydration of cinnamoyl-CoA, caffeoyl-CoA, and feruloyl-CoA to benzaldehyde, protocatechuic aldehyde, and vanillin, respectively (Fig. 5). These findings suggest the involvement of cinnamoyl-CoA hydratase/lyase in vanillin biosynthesis in *H. androsaemum* (Ahmed et al. 2002).

In another possibility, caffeic acid undergoes two consecutive methylations at the C4- and C3-positions of its aromatic ring to produce 3,4-dimethoxycinnamic acid (Fig. 5). Subsequently, this compound is demethylated and glycosylated before undergoing side chain shortening to produce vanillic acid or its glucoside (Funk and Brodelius 1990a, b), which are later reduced to vanillin and vanillin glucoside, respectively (Kundu 2017).

Recently, intense debate has emerged about the real participation of vanillin synthase (*Vp*VAN) in the direct conversion of ferulic acid to vanillin in *V. planifolia*, as proposed by Gallage et al. (2014). In this way, Yang et al. (2017) re-evaluated *Vp*VAN activity and verified that this protein acts as a 4-hydroxybenzaldehyde synthase. These authors affirmed that *Vp*VAN is identical at the sequence level to a protein identified in 2003 as being associated with the conversion of 4-coumaric acid to 4-hydroxybenzaldehyde (Havkin-Frenkel et al. 2003; Yang et al. 2017). However, the ability of *Vp*VAN to use ferulic acid as a substrate and, therefore, to act directly on vanillin biosynthesis has been reported by some independent studies (Havkin-Frenkel et al. 2006; Havkin-Frenkel and Podstolski 2007; Chee et al. 2017). According to Gallage et al. (2018), for the purification of *Vp*VAN protein, Yang and colleagues used an antibody prepared from a protein fraction capable of converting *p*-coumaric acid into *p*-hydroxybenzaldehyde; due to this, they did not observe *Vp*VAN activity on ferulic acid. In contrast, Gallage et al. (2014) purified the *Vp*VAN protein through a rabbit reticulocyte lysate-based transcription/translation system and demonstrated its in vitro activity on ferulic acid by highly sensitive LC–MS (ion trap) analyses. In addition, they did not observe any production of 4-hydroxybenzaldehyde when *p*-coumaric acid was used as the substrate (Gallage et al. 2014). Overall, the *Vp*VAN activity is

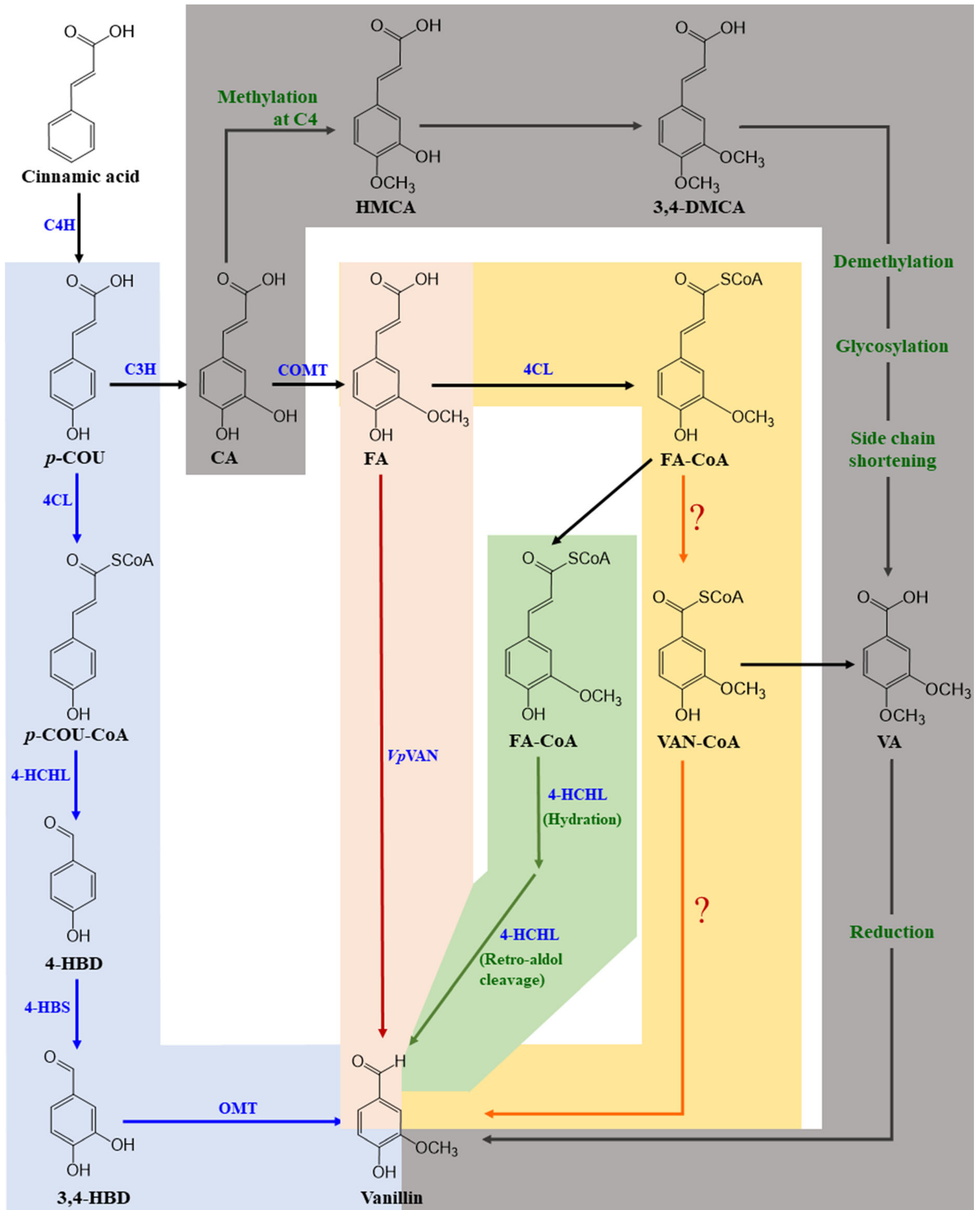
accepted now. It is important to note that the high identity in the amino acid sequence shared between *Vp*VAN and 4-hydroxybenzaldehyde synthase indicates that both enzymes are encoded by the same gene or are different isoforms, suggesting a common genetic origin (Yang et al. 2017).

Biosynthesis of salicylic acid

Salicylic acid is widely known for its pharmacological properties (Verberne et al. 2000; Wildermuth 2006; Zhang et al. 2013; Dieryckx et al. 2015). Abundant in plant species such as willow, myrtle, poplar, and meadowsweet, it has been used to relieve pain caused by several diseases since the fourth century B.C. However, it was isolated from the bark of willow only in the nineteenth century (Pierpoint 1997). Salicylic acid is a ubiquitous phytohormone that influences the response of plants under abiotic stresses such as drought, salinity and heavy metals (Veach et al. 2019; Sharma et al. 2020). Furthermore, salicylic acid retards petal senescence and induces flowering because it acts as an inhibitor of an ethylene-forming-enzyme, i.e. 1-aminocyclopropane-1-carboxylic acid (ACC) oxidase, which converts ACC to ethylene (Huang et al. 1993; Martínez et al. 2004; El-Tayeb 2005; Bahrami et al. 2013; Ji et al. 2016; Smith 2019). Salicylic acid also regulates thermogenesis in some plant species of the Araceae family (Raskin et al. 1987, 1989). It increases the expression of mitochondrial alternative oxidase and diverts electron flow from the cytochrome oxidase pathway to the cyanide-insensitive non-phosphorylating electron transport pathway (Raskin et al. 1987, 1989; Norman et al. 2004; Polidoros et al. 2005; Saha et al. 2016). In this way, some energy from electron transport is released as heat, which is used to raise the spadix temperature and volatilize pungent amines and indoles that attract pollinator insects. Salicylic acid is also a signal for the induction of localized and systemic acquired resistance (SAR) (Mustafa and Verpoorte 2005; Mustafa et al. 2009). SAR is characterized by reduced spread of the pathogen in the plant, in which pathogen resistance is observed not only close to the infected site but also in plant tissues distant from the infection point. In SAR, the accumulation of salicylic acid is accompanied by the biosynthesis of pathogenesis-related proteins (Durrant and Dong 2004; Mustafa and Verpoorte 2005; Pieterse et al. 2014).

Genetic evidence (Chen et al. 2009) suggests that more than 90% of the salicylic acid is formed from isochorismate (Fig. 3, yellow background), with the remaining salicylic acid biosynthesized from the cinnamic acid produced in the phenylpropanoid pathway by phenylalanine ammonia-lyase (PAL) (Fig. 3). For example, *A. thaliana* mutants that are deficient in an isoenzyme of ICS (*ics 1*) accumulate only 5–10% of the salicylic acid observed in wild plants infected by the virulent biotroph *Erysiphe* spp. or avirulent strains of *Pseudomonas syringae* (Wildermuth et al. 2001). Also, the salicylic acid content increased more than tenfold in control plants of *Nicotiana benthamiana* after 3 h of UV irradiation, but only fourfold in plants with silenced expression of ICS (Catinot et al. 2008). As isochorismate is converted to salicylic acid, this is an issue that needs be answered (Fig. 3, yellow background). In many bacteria, such as *Pseudomonas aeruginosa*, an isochorismate pyruvate lyase (IPL) catalyzes the production of salicylic acid and pyruvate by eliminating the enolpyruvyl moiety of isochorismate (DeClue et al. 2005; Martí et al. 2009). However, to date, no IPL has been identified in plants, suggesting that the biosynthesis of salicylic acid from isochorismate can be performed by a reaction different from that observed in bacteria (Chen et al. 2009). Nevertheless, the existence of an isochorismate pathway for salicylic acid biosynthesis in plants was firstly postulated by Verberne et al. (2000). In that study, the authors demonstrated that *N. tabacum* plants transformed with the bacterial ICS and IPL genes presented a constitutive accumulation of salicylic acid and an increase in resistance to various pathogens, in a similar manner to SAR.

Biochemical evidence supports the biosynthesis of salicylic acid from isochorismate (Fig. 3, yellow background). Accumulation of salicylic acid and 2,3-dihydroxybenzoic acid (2,3-DHBA) was accompanied by enhanced activity of ICS in *Catharanthus roseus* cell suspension cultures after elicitation with fungal cell-wall preparations (Moreno et al. 1996; Budi Muljono et al. 2002). In addition, the *C. roseus* cell suspension elicited with *Pythium aphanidermatum* extract and fed with [1-^{13}C]-D-glucose revealed a labeling profile in salicylic acid and 2,3-DHBA which is compatible with their biosynthesis from the isochorismate pathway (Mustafa et al. 2009). The authors also showed that the biosynthesis of salicylic acid precedes 2,3-DHBA. This finding was attributed to the



Cinnamic acid
C4H
p-COU
C3H
CA
COMT
FA
4CL
FA-CoA
Methylation at C4
HMCA
3,4-DMCA
Demethylation
Glycosylation
Side chain shortening
4CL
p-COU-CoA
4-HCHL
4-HBD
4-HBS
3,4-HBD
OMT
VpVAN
?
FA-CoA
4-HCHL (Hydration)
4-HCHL (Retro-aldol cleavage)
VAN-CoA
VA
Reduction
Vanillin

◀**Fig. 5** Biosynthesis of vanillin in plants. Vanillin may be formed by at least five distinct metabolic pathways, which occur in plastids or in re-differentiated plastids (phenyloplasts). On all routes, metabolites of the phenylpropanoid pathway act as precursors. In the CoA-independent non-β-oxidative pathway (pink background), which occurs in *V. planifolia*, a vanillin synthase (*Vp*VAN) converts ferulic acid (FA) to vanillin. In another CoA-independent pathway (gray background), caffeic acid (CA) undergoes several reactions to produce vanillic acid (VA), which is reduced to vanillin. Vanillin can also be produced from feruloyl-CoA (FA-CoA) via a two-step reaction catalyzed by 4-hydroxycinnamoyl-CoA hydratase/lyase (4-HCHL) (green background). In this CoA-dependent non-β-oxidative pathway, FA-CoA is first hydroxylated before undergoing a retro-aldol cleavage. Another CoA-dependent non-β-oxidative pathway (blue background), which uses *p*-coumaric acid (*p*-COU) as a precursor is still possible. In this route, *p*-COU is successively esterified with CoA, converted to 4-hydroxybenzaldehyde (4-HBD), hydroxylated to 3,4-dihydroxybenzaldehyde (3,4-HBD) and methylated to produce vanillin. Finally, FA can be successively converted to feruloyl-CoA (FA-CoA), vanilloyl-CoA (VAN-CoA) and then vanillin (yellow background). The enzymes involved in this CoA-dependent β-oxidative pathway are unknown. Cinnamate 4-hydroxylase (C4H); *p*-coumarate 3-hydroxylase (C3H); *p*-coumarate-CoA: ligase (4CL); 4-hydroxybenzaldehyde synthase (4-HBS); *O*-methyltransferase (OMT); cinnamyl (caffeate) *O*-methyltransferase (COMT); 3-hydroxy-4-methoxycinnamic acid (HMCA); 3,4-dimethoxycinnamic acid (3,4-DMCA)

fact that salicylic acid is a signal compound in the SAR response that culminates with the production of other compounds, including 2,3-DHBA (Mustafa et al. 2009).

After biosynthesis in the plastid, the transport of salicylic acid to the cytosol is performed by a carrier called Enhanced Disease Susceptibility 5 (EDS5) (Fig. 3), a homologous protein with members of the multidrug and toxin extrusion transporter family (Ishihara et al. 2008; Chandran et al. 2014). Its expression in *A. thaliana* is induced by pathogens, UV-C light and SA (Nawrath 2002). In the cytosol, salicylic acid can be metabolized to 2,3-DHBA and 2,5-DHBA (gentisic acid) (Fig. 3), which may be conjugated to sugars due to their toxicity (Bartsch et al. 2010; Huang et al. 2018). It is possible that 2,5-DHBA exerts a complementary role to that of salicylic acid, since it induces the expression of pathogenesis-related genes not induced by salicylic acid (Bellés et al. 1999). Initially, it was thought that 2,3-DHBA and 2,5-DHBA originated from the non-enzymatic reaction of salicylic acid with hydroxyl radical ($HO^{\cdot -}$). However, two hydroxylases have recently been identified in *A. thaliana*, both of which show expression induced by salicylic acid (Zhang et al. 2013, 2017). In vitro, salicylic acid 3-hydroxylase (S3H) can hydroxylate salicylic acid at C3 and C5 to produce, respectively, 2,3-DHBA and 2,5-DHBA (Fig. 3), although only C3-hydroxylase activity has been observed in vivo (Zhang et al. 2013). The salicylic acid 5-hydroxylase catalyzes the production of 2,5-DHBA in vivo and exhibits inhibition by the substrate. S5H also has a higher affinity (Km = 5.15 μM) and specificity constant (Kcat/Km = 4.96×10^4 M^{-1} s^{-1}) for salicylic acid than S3H (Km = 58.29 μM and Kcat/Km = 6.09×10^3 M^{-1} s^{-1}), emphasizing its primordial role in the catabolism of this phytohormone (Zhang et al. 2017). In addition, S3H is expressed primarily at stages of senescence, whereas S5H, as well as being constitutively expressed, may have increased expression at the senescence stage. Together, the biochemical properties of S5H enable it to perform the fine control of salicylic acid levels during the development of *A. thaliana*. *Escherichia coli* has a distinct pathway for producing 2,3-DHBA and 2,5-DHBA, which uses a 2,3-dihydro-2,3-dihydroxybenzoate synthase and 2,3-dihydro-2,3-dihydroxybenzoate dehydrogenase (Khalil and Pawelek 2011). In plants, enzymes homologous to these act as nicotinamidases and are involved in the salvage pathway of NAD (Hunt et al. 2007).

As previously described, the biosynthesis of salicylic acid from *t*-cinnamic acid is possible and involves the CoA-dependent β-oxidative, CoA-independent non-oxidative and CoA-dependent non-oxidative pathways (Fig. 3). In some cases, the benzoic acid acts as an intermediate because the shortening of the propanoid side chain of *t*-cinnamic acid precedes the hydroxylation of its aromatic ring (Fig. 3, blue background) (León et al. 1995; Chen et al. 2009). After the conversion of *t*-cinnamic acid to benzoic acid, a monooxygenase P450 enzyme called benzoic acid 2-hydroxylase (BA2H) (León et al. 1995), whose expression is induced by tobacco mosaic virus, hydrogen peroxide or benzoic acid (Dempsey et al. 2011), catalyzes the hydroxylation of benzoic acid to produce salicylic acid (León et al. 1995). However, in elicited *N. tabacum* cells, the accumulation of salicylic acid involves de novo biosynthesis of BA and had benzoyl–glucose as the main intermediate, instead of

free BA (Chong et al. 2001). The production of salicylic acid from *t*-cinnamic acid, via BA, participates in the stress-induced flowering in *Pharbitis nil* (Hatayama and Takeno 2003). Also, the non-oxidative pathway is not present in *Cucumis sativus* and *Nicotiana attenuata* (Jarvis et al. 2000; Mustafa and Verpoorte 2005). In *Primula acaulis* and *Gaultheria procumbens*, salicylic acid is produced via *o*-coumaric acid, since hydroxylation of the aromatic ring precedes the reactions of β-oxidation (Fig. 3) (Chen et al. 2009).

The role of salicylic acid as a signal for SAR was first demonstrated in *N. tabacum* plants, after the injection of salicylic acid into their leaves. These plants exhibited an accumulation of pathogenesis-related proteins and enhanced resistance to the tobacco mosaic virus (White 1979; Antoniw and White 1980). Early studies indicated salicylic acid as a signaling molecule produced at the infected site and subsequently transported to uninfected plant tissues to induce SAR. This hypothesis was supported by the finding that salicylic acid-deficient *N. tabacum* plants, expressing a salicylate hydroxylase (nahG) gene from *Pseudomonas putida,* displayed suppressed SAR and the expression of systemic pathogenesis-related genes (Gaffney et al. 1993; Delaney et al. 1994; Vernooij et al. 1994). However, several grafting experiments showed that, in *N. tabacum*, methyl salicylate, but not salicylic acid, is the mobile signal. In this way, the SAR response was induced in the wild scion grafted onto salicylic acid-deficient rootstock of transgenic *N. tabacum* plants (Vernooij et al. 1994). However, salicylic acid accumulation has been shown to be crucial for SAR induction, which has been fully suppressed in plants with a salicylic acid-deficient scion grafted on to wild rootstock (Vernooij et al. 1994). In addition, the presence of salicylic acid methyltransferase 1 (SAMT1), an enzyme that converts salicylic acid to methyl salicylate, is required in tissues that produce the mobile signal. Also, the methyl salicylate esterase activity of salicylic acid-binding protein 2 (SABP2), which converts methyl salicylate to salicylic acid, is necessary for signal perception in systemic tissues of *N. tabacum* (Forouhar et al. 2005; Park et al. 2007). Although methyl salicylate is the mobile signal for SAR induction in *N. tabacum* (Hayat et al. 2010; Dieryckx et al. 2015; Hammerbacher et al. 2019), it is yet possible to know whether it plays a similar role in other plant species. It is important to note that the methyl group makes salicylic acid a volatile molecule. In fact, methyl salicylate is carried by the phloem from infected tissues to non-infected tissues, but may also act as a volatile signal able to induce SAR response in neighboring plants (Hammerbacher et al. 2019).

In addition to its role in communicating with other plants, methyl salicylate may participate in the communication of plants with organisms of the second (herbivores and pollinators) and third (enemies of herbivores) trophic level (Holopainen and Blande 2012). For instance, the methyl salicylate signal emitted by pathogen-infected plants, or plants damaged by arthropod pests, can be recognized by parasitoid insects to identify their prey (De Boer and Dicke 2004; James and Price 2004; Mallinger et al. 2011; Holopainen and Blande 2012; Stepanycheva et al. 2016; Hammerbacher et al. 2019). In this way, *Candidatus Liberibacter asiaticus* (Las) alters the odors released by citrus trees to attract its vector *Diaphorina citri*. Additionally, the emission of methyl salicylate by an infected plant attracts the parasitoid of *D. citri*, *Tamarixia radiata* (Martini et al. 2014). On the other hand, Snoeren et al. (2010) demonstrated that *Piere rapae* infested *AtBSMT1-KO* mutant *A. thaliana* plants, deficient in methyl salicylate biosynthesis, were more attractive to the parasitoid *Diadegma semiclausum* than wild plants.

Plant roots can exude a variety of compounds such as sugars, amino acids, organic acids and phenolic compounds that modify the physical properties of the soil, act as allelopathic and herbivorous defense agents, and regulate the microbial community in the rhizosphere (Rasmann and Turlings 2016; Venturi and Keel 2016). The rhizosphere is constituted by rhizobial bacteria, mycorrhizal fungi, plant growth-promoting bacteria and fungi, as well as nematodes. All these organisms may eventually make beneficial mutualistic associations with plants because they can release antibiotics and/or secondary metabolites capable of inhibiting pathogen proliferation; they also elicit induced systemic resistance (ISR) in host plants (Lugtenberg and Kamilova 2009; Kobayashi 2015). Therefore, the ability of plants to influence the rhizomicrobiome may contribute to plant growth and development. However, only some of the substances used by plants to modify rhizomicrobiome are known (Venturi and Keel 2016). Several recent studies have strikingly shown that salicylic acid and methyl

salicylate act as modulators of colonization of the root microbiome (Doornbos et al. 2011; Kobayashi 2015; Lebeis et al. 2015; Igiehon and Babalola 2018; Veach et al. 2019). For a detailed discussion on plant-microorganism and microorganism-plant signaling in the rhizosphere, see Venturi and Keel (2016).

Metabolic actions of simple phenolic acids on plants

Simple phenolic acids are among the most widely distributed phenolic compounds in plants and they play important biological roles. Many of them, such as hydroxycinnamic acids, are useful in the biosynthesis of structural components of the cell wall (de Oliveira et al. 2015). Others are crucial for defense responses to pathogens and herbivores (as described earlier) or act as signaling molecules in the establishment of legume-rhizobia or arbuscular mycorrhizal symbioses (Mandal et al. 2010). While flavonoids have a well-defined role as a signaling molecule in plant–microbe interactions, the function of phenolic acids in this process is not clear. As an example, the protocatechuic acid, 4-hydroxybenzaldehyde and *p*-coumaric acid present in the root nodules of *Vigna mungo* stimulate IAA production and regulate the morphogenesis of its symbiont *Rhizobium* spp. (Mandal et al. 2009). However, the mechanism by which the infection efficiency is improved by these phenolic acids needs to be established. In addition, the expression of nod genes can be stimulated or inhibited by phenolic acids; a property that appears to depend on their chemical structure and concentration, and to be strain-specific (Djordjevic and Rolfe 1988; Seneviratne and Jayasinghearachchi 2003). Also, phenolic acids have antioxidant activity comparable to that of antioxidant vitamins (Tsao and Deng 2004), besides conferring to foods characteristics such as bitterness, astringency, color, taste, odor, and stability against lipid oxidation (Chandrasekara 2019). More pertinent to the purposes of this review, however, are the phytotoxic effects of phenolic acids on plants, as well as the allelopathic effects resulting from their release into the environment.

Simple phenolic acids are the most common and important class of allelochemicals present in ecosystems (Li et al. 2010). They can be released into the environment through root exudation or by leaching of roots, aerial part of plants or decomposing plant residues (Fig. 2). They are important precursors of humic substances in soils. Considering the total amount of phenolic compounds in plants and organic matter it is estimated that more than 100 kg of these compounds are released per hectare of pasture annually (Tharayil et al. 2006). Previous studies have reported that its contents in rhizosphere and soils can reach 90 ppm (Whitehead 1964). Among these numerous phenolic acids, the most common are benzoic acids and its derivatives (salicylic, *p*-hydroxybenzoic, gallic and protocatechuic acids) and cinnamic acid and its derivatives (*p*-coumaric, caffeic, ferulic and sinapic acids) (Fig. 1). All these phenolic acids were identified in root exudates, leaf leachates or the residues of decomposing plants.

Although phenolic acids can inhibit the growth of many plants in laboratory experiments, their allelopathic actions in soils remain a controversial matter. Inderjit and Bhowmik (2004) did not observe any allelopathic effect on *C. sativus* and *Raphanus sativus* when cultivated on soil containing 1 to 20 mg L^{-1} of benzoic acid. This is because simple phenolic acids are rapidly metabolized by microorganisms (Li et al. 2010). Some authors argue that the allelopathic effects of phenolic acids on the environment are unlikely, even though few data support this hypothesis (Blum 2004). Due to their high biodegradability, the allelopathic effects (it perhaps being more acceptable to call them phytotoxic effects) of phenolic compounds are investigated using, in many cases, artificial means of cultivation such as hydroponics, agar or sand (Inderjit and Bhowmik 2004).

Another factor that negatively contributes to the allelopathic effect of phenolic acids in the environment is their adsorption by soil particles. Phenolic acids may bind to soil particles through van der Waals interactions or hydrogen bonds, but may be released as a result of the decrease in soil solution strength or the presence of competing ions (Dalton 1999). The ionization state of the carboxyl group of phenolic acids can vary with pH, which plays an important role in the adsorption of these compounds by the soil particles. Thus, at pH lower than 4.5, the phenolic acids are in the non-ionized (protonated) form and are adsorbed by organic matter and soil clay. In contrast, at slightly acidic or basic pH, the phenolic acids are negatively charged and are adsorbed by the colloids (positively charged) of the soil. In addition, polyvalent

cations can function as bridges in the interaction between negatively charged phenolic acids and soil particles containing negative charges (Greeland 1971).

Although the phytotoxic effects of phenolic acids have been extensively investigated, few studies have even identified their primary molecular targets. In fact, since 2007 more than 1700 articles have been published on allelopathy, allelochemicals, and phytotoxicity; 41% of which cover simple phenolic acids (Macías et al. 2019). However, as has been so well pointed out by these authors, the use and promising applications of phenolic acids may have been overestimated by researchers. Moreover, studies are lacking which assess whether structurally similar phenolics act on the same primary target. Indeed, identifying the primary molecular target for allelochemical or natural products is a challenging task because they commonly have wide actions, i.e., they can act on more than one target simultaneously. Furthermore, if the onset of symptoms in plants is not immediate, secondary effects can be confused with primary effects (Duke 1985). Nevertheless, the identification of the primary molecular target of phenolic acids is crucial to the understanding of their ecological role as well as being important in the search for new mechanisms of action of herbicides (Trezzi et al. 2016). From the findings reported, and described in detail later, it is reasonable to assume that phenolic acids act primarily at the plasma membrane level, reducing electrical conductivity and nutrient uptake by the roots. Additionally, primary effects on mitochondria and chloroplasts have also been reported from studies using in vitro approaches. Secondarily, effects on photosynthesis, cellular respiration, hormonal balance (mainly with abscisic, indole-3-acetic and gibberellic acids, and ethylene), stomatal closure and carbon allocation to the roots have been noted (Einhellig 2004). Tertiary effects have also been described and are long-term effects resulting from a primary event. In brief, the consequence of the phytotoxic effects of phenolic acids is the reduction of plant growth and development. Next, we describe the primary and secondary effects of simple phenolic acids on plants, as summarized in Table 1.

Primary effects of simple phenolic acids on plants

Plasma membrane

The plant root is the first organ that meets phenolic acids present in soils. Consequently, it constitutes the first site of action for these compounds. Among the processes involved are the absorption of water and nutrients. There is evidence that the absorption of phenolic acids can reduce the uptake of nutrients, which is directly related to the fact that their absorption is dependent on pH. In principle, the entry of phenolic acids into the cells dissipates the proton gradient across the membrane, essential for the transport of ions to the cellular interior (Fig. 6). The cell exterior (apoplast) is a slightly more acidic environment than the inside of the cells. Part of the phenolic acids is protonated at acidic pH. In this condition, they are hydrophobic and pass freely through the plasma membrane. Upon entering the cells, they find a higher pH and then dissociate, dissipating the H^+ gradient (Lee 1977). This mechanism was confirmed for benzoic and cinnamic acids more than three decades ago (Chipley 2005).

The mechanism by which phenolic acids influence nutrient absorption is not restricted to the ability to depolarize membranes (Fig. 6). For example, benzoic and cinnamic acids also induce a leakage of UV light-absorbing compounds by roots of *Glycine max* seedlings grown in hydroponics (Baziramakenga et al. 1995). The leakage of UV light-absorbing compounds indicates the loss of membrane integrity. The process is also accompanied by the degradation of lipids and proteins of membrane. The depolarization of membrane is accompanied by rapid increase in the production of ROS. Consequently, lipid peroxidation increases and the activities of ROS-scavenging enzymes, such as peroxidase (POD) and catalase (CAT) (Baziramakenga et al. 1995). These researchers also observed a reduction in the number of sulfhydryl groups, after treatments of *G. max* seedlings with phenolic acids. This indicates that these allelochemicals cause, directly or indirectly, the oxidation of these chemical groups, which are important constituents of the ATPases enzymes, inducing the formation of disulfide bonds. Carriers, pumps and ion channels, fundamental for the maintenance of the membrane potential or the proton gradient through the membrane, can be affected by this oxidative process,

Table 1 Summary of some physiological/biochemical effect of simple phenolic acids on plants

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Effects on plasma membrane			
Benzoic acid	–	Dissipates the proton gradient across the membrane	Chipley (2005)
	Glycine max	Loss of membrane integrity. Degradation of lipids and proteins of membrane	Baziramakenga et al. (1995)
Salicylic acid	*Solanum lycopersicum*	Tolerance to salinity. Higher stem growth rate under saline stress. Protection of membrane integrity	Stevens et al. (2006)
Cinnamic acid	–	Dissipates the proton gradient across the membrane	Chipley (2005)
	G. max	Loss of membrane integrity. Degradation of lipids and proteins of membrane	Baziramakenga et al. (1995)
Ferulic and vanillic acids	*Cucumis sativus*	Inhibit phosphorus uptake	Lyu et al. (1990)
Effects on water relations			
p-Hydroxybenzoic acid	*G. max*	Reduction of leaf water potential	Barkosky and Einhellig (2003)
Cinnamic acid	*Lolium perenne, Dactylis glomerata* and *Rumex acetosa*	Reduce the relative water content (RWC). Inhibition of growth and reduction of the leaf water potential	Hussain and Reigosa (2011)
	Lactuca sativa	Applied together with ABA caused additive inhibitory effect on growth	Li et al. (1993)
Caffeic acid	*Euphorbia esula*	Inhibition of growth and changes in water relations	Barkosky et al. (2000)
	C. sativus	Pretreatment increase the tolerance of seedlings to dehydration	Wan et al. (2014)
Effects on hormonal balance			
Benzoic acid	*Pyrus communis*	Inhibition of ethylene production at high concentration	Leslie and Romani (1988)
	S. lycopersicum and *Malus domestica*	Inhibition of ethylene production	Baker et al. (1978)
Gallic acid	*Malus* 'Jork 9'	Increase the rooting and massive inhibition of decarboxylation of IAA	de Klerk et al. (2011)
Syringic and protocatechuic acids	*Vicia faba*	Restored the stomatal closure induced by ABA	Purohit et al. (1991)
Gentisic and vanillic acids	*P. communis*	Inhibition of ethylene production from 1-aminocyclopropane-1-carboxylate (ACC)	Leslie and Romani (1988)
Tannic acid, fluoroglucinol and pyrogallol	*Malus* 'Jork 9'	Increase the rooting and massive inhibition of decarboxylation of IAA	de Klerk et al. (2011)
Salicylic acid	*V. faba*	Restored the stomatal closure induced by ABA	Purohit et al. (1991)
	P. communis	Strong inhibition of ethylene production from 1-aminocyclopropane-1-carboxylate (ACC)	Leslie and Romani (1988)
Salicyl alcohol, salicin, *p*-amino salicylate and 3,5-dinitrosalicylate derivatives	*P. communis*	Inhibition of ethylene production from 1-aminocyclopropane-1-carboxylate (ACC)	Leslie and Romani (1988)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Cinnamic acid	*G. max*	Increase the activity of IAA oxidase in roots	Salvador et al. (2013)
	Pisum sativum	Antagonized the growth-promoting effects of IAA	Van Overbeek et al. (1951)
p-Coumaric acid	*L. sativa*	Applied together with ABA caused additive inhibitory effect on growth	Li et al. (1993)
Caffeic acid	*V. faba*	Reversed ABA effect	Purohit et al. (1991)
	L. sativa	Reversed the growth inhibition caused by ABA	Li et al. (1993)
Ferulic acid	*Malus* 'Jork 9'	Increase the rooting and massive inhibition of decarboxylation of IAA	de Klerk et al. (2011)
	Zea mays	Increase activity of IAA oxidase in seedlings	Devi and Prasad (1996)
	Oryza sativa	Overexpression of genes involved in the biosynthesis of jasmonic and ethylene; inhibition of formation of lateral roots and root hairs	Chi et al. (2013)
	L. sativa	Reversed the growth inhibition caused by ABA	Li et al. (1993)
	V. faba	Reversed ABA effect	Purohit et al. (1991)
Vanillin, catechol and chlorogenic acid	*Malus* 'Jork 9'	Increase the rooting and massive inhibition of decarboxylation of IAA	de Klerk et al. (2011)
Effects associated with oxidative stress and enzyme activities			
Benzoic acid	*G. max*	Reductions in the activities of ROS-scavenging enzymes, such as peroxidase (POD) and catalase (CAT)	Baziramakenga et al. (1995)
	C. sativus	Stimulus of activities of SOD and POD	Yu et al. (2003)
Phenylpropionic acid	*C. sativus*	Stimulus of activities of SOD and POD	Yu et al. (2003)
o-Hydroxybenzoic acid			
p-Hydroxybenzoic acid			
3,4-Dihydroxybenzoic acid			
Vanillic acid			
Gallic acid	*O. sativa*	Minimized the damage caused by NaCl or polyethylene glycol stresses. Reduction of lipid peroxidation and increase of SOD, POD and APX activities	Ozfidan-Konakci et al. (2015)
	C. sativus	Stimulus of activities of SOD and POD	Baziramakenga et al. (1995) Yu et al. (2003)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Cinnamic acid	*G. max*	Reductions in the activities of ROS-scavenging enzymes, such as POD and CAT	Baziramakenga et al. (1995)
	C. sativus	Stimulus of activities of SOD and POD	Yu et al. (2003)
	C. sativus	Increased the ability of plants to resist water stress. Decrease of H_2O_2 and MDA contents in PEG treated plants. activation of POD, CAT, GR, GPX and DHAR. Accumulation of free sugars in leaves	Sun et al. (2012)
	C. sativus	Activation of SOD, CAT, POD and NADH oxidase. increase of $O_2^{\cdot -}$ and H_2O_2 contents	Ding et al. (2007a, b)
	Z. mays	Reduce the oxidative damage under salt stress. Increase of SOD activity. Reduce CAT activity and MDA content in leaves	Singh et al. (2013)
	G. max	Decrease the H_2O_2 content in roots. Increase of soluble and cell wall-bound POD activities and lignin content	Bubna et al. (2011)
p-Coumaric acid	*C. sativus*	Increase in SOD and POD activities	Yu et al. (2003)
Caffeic acid	*C. sativus*	Increase in SOD, POD, GPX, APX and MDAR activities	Wan et al. (2014)
	C. sativus	Stimulus of activities of SOD and POD	Yu et al. (2003)
	Vigna radiata	Activation of SOD, CAT, guaiacol peroxidase, and GR. Increase of H_2O_2 and MDA contents	Singh et al. (2009)
Hydrocaffeic acid	*C. sativus*	Increase in SOD and POD activities	Yu et al. (2003)
Ferulic acid	*O. sativa*	Increase level of ROS, lipid peroxidation and intracellular calcium. Activation of lipoxygenase	Chi et al. (2013)
	C. sativus	Increase in SOD and POD activities	Yu et al. (2003)
Vanillin	*S. lycopersicum*	Stimulus in activities of POD, SOD CAT and PPO	Ghareib et al. (2010)
	O. sativa	Dose-dependent stimulus of POD activity	Jazayeri et al. (2007)
Sinapic acid	*C. sativus*	Stimulus of activities of SOD and POD	Yu et al. (2003)
Effects on photosynthesis			
Benzoic acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
	G. max	Increase of photosynthetic rate and stomatal conductance in plants under drought stress	Anjum et al. (2013)
Phenylpropionic acid	*C. sativus*	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
o-hydroxybenzoic acid	*C. sativus*	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
p-Hydroxybenzoic acid	*G. max*	Stomatal closure	Barkosky and Einhellig (2003)
	G. max	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
	Echinochloa crus-galli	Reduce the chlorophyll content	Esmaeili et al. (2012)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
	L. sativa	Reduction of Fv/Fm, photochemical fluorescence quenching and non-photochemical fluorescence quenching. Lower ratios of intercellular CO_2 concentration from leaf to air and carbon isotope discrimination values	Hussain et al. (2010)
3,4-Dyhydroxybenzoic acid	*C. sativus*	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
O-Hydroxyphenylacetic acid	*O. sativa*	Reduce the content of chlorophyll and three of its biosynthetic precursors, protoporphyrin IX, Mg-protoporphyrin and protochlorophyllide; Stimulus of chlorophyllase and Mg-dequelatase activities	Yang et al. (2002, 2004)
Vanillic acid	*E. crus-galli*	Reduce the chlorophyll content	Esmaeili et al. (2012)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
Gallic acid	*V. faba*	Restored the stomatal closure induced by ABA	Purohit et al. (1991)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
Syringic acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
Protocatechuic acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
Gentisic acid	*G. max* and *Z. mays*	Stimulus of photosynthetic rate, stomatal conductance and transpiration	Khan et al. (2003)
Tannic acid	*V. faba*	Restored the stomatal closure induced by ABA	Purohit et al. (1991)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Salicylic acid	*G. max* and *Z. mays*	Stimulus of photosynthetic rate, stomatal conductance and transpiration	Khan et al. (2003)
	Z. mays	Stimulus of photosynthetic rate, stomatal conductance and transpiration	Zhou et al. (1999)
	Brassica juncea	Increase the photosynthetic rate, stomatal conductance and transpiration	Fariduddin et al. (2003)
	S. lycopersicum	Induced tolerance to salinity. Increase of photosynthetic rate, stomatal conductance and transpiration	Stevens et al. (2006)
Acetylsalicylic acid	*G. max* and *Z. mays*	Stimulus of photosynthetic rate, stomatal conductance and transpiration	Khan et al. (2003)
Cinnamic acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994, Einhellig and Rasmussen (1979), Patterson (1981)
	E. crus-galli	Reduce the chlorophyll content	Esmaeili et al. (2012)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
	L. perenne, D. glomerata and *R. acetosa*	Reduce the Fv/Fm and Φ_{PSII}. Induce the stomatal closure	Hussain and Reigosa (2011)
	V. unguiculata	Decrease of photosynthetic rate and rubisco activity. No effect on chlorophyll fluorescence and chlorophyll content	Huang and Bie (2010)
	C. sativus	Decrease of photosynthetic rate, stomatal conductance and Φ_{PSII}	Ye et al. (2004)
p-Coumaric acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
	O. sativa	Reduce the content of chlorophyll and three of its biosynthetic precursors, protoporphyrin IX, Mg-protoporphyrin and protochlorophyllide. Stimulus of chlorophyllase and Mg-dequelatase activities	Yang et al. (2002, 2004)
	E. crus-galli	Reduce the chlorophyll content	Esmaeili et al. (2012)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
Caffeic acid	*E. esula*	Stomatal closure and lower transpiration rates	Barkosky et al. (2000)
	G. max	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
Hydrocaffeic acid	*C. sativus*	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Ferulic acid	*G. max*	Reduce the chlorophyll content	Baziramakenga et al. (1994), Einhellig and Rasmussen (1979), Patterson (1981)
	O. sativa	Reduce the content of chlorophyll and three of its biosynthetic precursors, protoporphyrin IX, Mg-protoporphyrin and protochlorophyllide; Stimulus of chlorophyllase and Mg-dequelatase activities	Yang et al. (2002, 2004)
	E. crus-galli	Reduce the chlorophyll content	Esmaeili et al. (2012)
	C. sativus	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
	L. sativa	Reduction of Fv/Fm, photochemical fluorescence quenching and non-photochemical fluorescence quenching. Lower ratios of intercellular CO_2 concentration from leaf to air and carbon isotope discrimination values	Hussain et al. (2010)
	Several crops	Reduce stomatal conductance and photosynthesis	Einhellig (1996)
Sinapic acid	*C. sativus*	Reduce the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration	Yu et al. (2003)
Juglone	*Lemna minor*	Reduction of chlorophyll content, net photosynthesis	Hejl et al. (1993)
	G. max	Reduction of photosynthesis	Hejl et al. (1993)
	P. sativum	Inhibition of oxygen evolution by chloroplasts	Hejl et al. (1993)
Phthalic and syringic acids	Strawberry	(*Fragaria* × *ananassa* Duch.)	At low concentration, stimulus of biosynthesis of photosynthetic pigments, stomatal conductance and photosynthetic rate
			At high concentration, reduction of photochemical quenching coefficient and primary light energy conversion by PSII and electron transport rate. ATP synthesis and carbon assimilation impaired
Lu et al. (2018)			
Effects on respiration			
Vanillic acid	*Solanum tuberosum*	Inhibition of activity of succinate dehydrogenase and respiration	Makovec and Sindelár (1984)
Gallic acid		Inhibition of succinate dehydrogenase, malate dehydrogenase, cytochrome *c* oxidase activities and respiration	Makovec and Sindelár (1984)
Salicylic acid	*Beta vulgaris*	Reduction of respiratory activity	Kluge et al. (2010)
Protocatechuic acid	*Phaseolus aureus*	Inhibition of respiration in isolated mitochondria	Demos et al. (1975)

Table 1 continued

Simple phenolic acid	Plant species	Physiological/biochemical effects	References
Cinnamic acid	*S. tuberosum*	Inhibition of activity of succinate dehydrogenase and respiration	Makovec and Sindelár (1984)
	G. max	Inhibition of rate oxygen consumption in cotyledons	Peñuelas et al. (1996)
	Chlorella vulgaris	Stimulus in respiration	Dedonder and Van Sumere (1971)
p-Coumaric acid	*P. aureus*	Inhibition of respiration in isolated mitochondria	Demos et al. (1975)
m-Coumaric acid	*E. crus-galli*	Reduce the chlorophyll content	Esmaeili et al. (2012)
Caffeic acid	*P. aureus*	Inhibition of respiration in isolated mitochondria	Demos et al. (1975)
	S. tuberosum	Inhibition of activity of succinate dehydrogenase and respiration	Makovec and Sindelár (1984)
Chlorogenic acid	*S. tuberosum*	Inhibition of activity of succinate dehydrogenase and respiration	Makovec and Sindelár (1984)
Juglone	*G. max*	Stimulus of oxygen uptake in isolated mitochondria	Hejl et al. (1993)
Rosmarinic acid	*Arabidopsis thaliana*	Inhibition of CAT and POD activities and increase of ROS and malondialdehyde contents. Dissipation of mitochondrial transmembrane potential ($\Delta\Psi_m$). Reduction of metabolites of citric acid cycle and biosynthesis of related amino acid	Araniti et al. (2018)
Syringic and phthalic acids	Strawberry	(*Fragaria* × *ananassa* Duch.)	Increase of H_2O_2 and $O_2^{\cdot-}$ contents. Inhibition of POD and SOD activities. Increase in the degree of opening of the mitochondrial membrane permeability transition pore. Decrease of mitochondrial transmembrane potential ($\Delta\Psi_m$) and cytochrome c/a ratio
Wang et al. (2019)			

amplifying the proton gradient dissipation (Baziramakenga et al. 1995). The third level of action of phenolic acids is its influence on the activity of cellular receptors for hormones, thereby affecting the mechanisms of signal transduction across the membrane (Einhellig 2004).

Cellular respiration and photosynthesis

A limited number of in vitro studies have revealed that electron transport reactions in mitochondria and chloroplasts may be one of the primary targets for the action of phenolic acids. In general, cinnamic acid derivatives have more potent effects on mitochondria and chloroplasts than benzoic acid derivatives.

In mitochondria, phenolic acids act mainly as inhibitors of electron transport and ATP synthesis or as weak uncouplers. For instance, *p*-coumaric and gentisic acids induced loss of respiratory control and decreased the state III respiration rates in mitochondria isolated from *Phaseolus aureus* hypocotyls (Demos et al. 1975). For these authors, the loss of respiratory control did not occur due to the uncoupling activity of the phenolic acids because no stimulus of respiration was observed. Caffeic and protocatechuic acids reduced the respiration rate only at high concentrations (500 μM) and, then, without any physiological significance. The phenolic acids also reduced the phosphate-dependent uptake of Ca^{2+} by the mitochondria. Notably, vanillic acid selectively

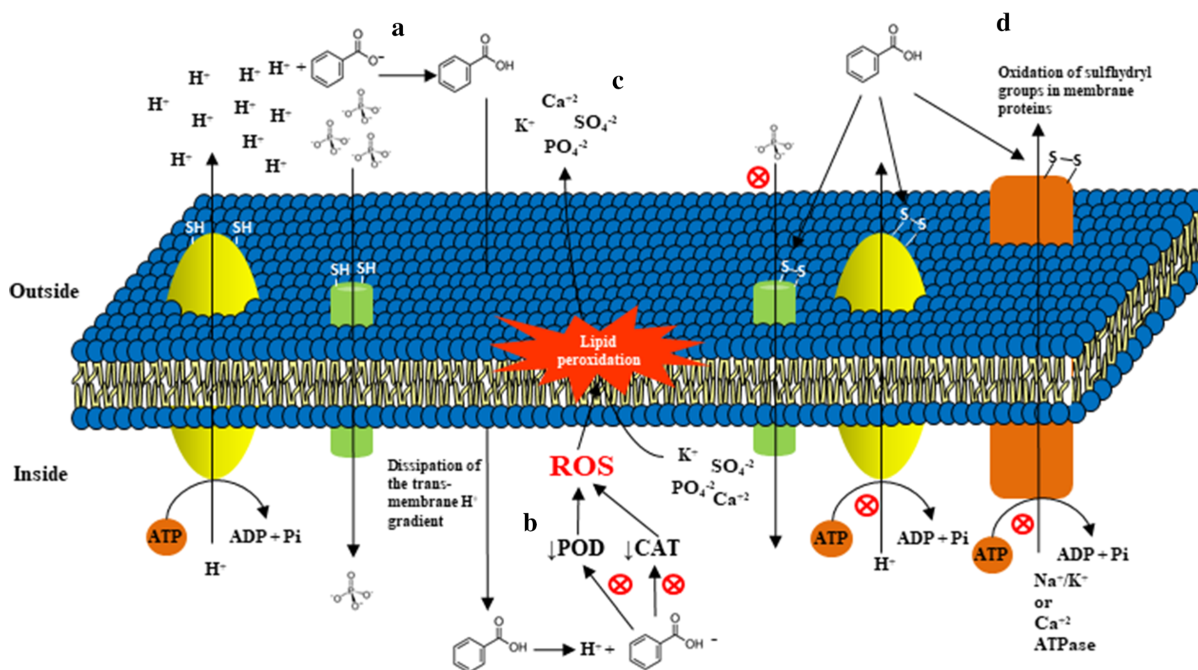


Fig. 6 Effects of simple phenolic acids on plasma membrane. The membranes of the root cells are the first site of action for simple phenolic acids. Initially, simple phenolic acids, such as benzoic acid, enter the cells and dissipate the transmembrane H^+ gradient (**a**). Within the cells, simple phenolic acids inhibit the activity of antioxidant enzymes such as peroxidase (POD) and catalase (CAT), leading to the increase of reactive oxygen species (ROS) and to lipid peroxidation (**b**). The degradation of lipids and proteins of membrane leads to depolarization of the plasma membrane due to the leakage of the K^+, Ca^{2+}, PO_4^{2-} and SO_4^{2-} ions (**c**). Single phenolic acids also inhibit the activity of membrane proteins (H^+, Ca^{2+} and Na^+/K^+ ATPases) through the oxidation of their sulfhydryl groups (**d**)

inhibited the uptake of Ca^{2+} without affecting respiration or respiratory control. In this study, also, the rate of reduction of externally-added cytochrome *c* was used to determine the integrity of the outer mitochondrial membrane. Only gentisic acid increased the rate of cytochrome *c* reduction and generated a loss of outer membrane integrity. All the phenolic acids that affected mitochondrial respiration reduced the length of *P. aureus* hypocotyls. Unsurprisingly, mitochondrial energy metabolism is crucial to seedling growth and development.

In another study performed with mitochondria isolated from *P. aureus* hypocotyls, cinnamic and benzoic acids acted as electron transport inhibitors; benzoic acid was the weaker (Moreland and Novitzky 1987). In coupled mitochondria, the phenolic acids inhibited the phosphorylation of ATP more intensely than the electron transport reactions. In addition, electron transport in uncoupled mitochondria was less sensitive. Therefore, the main inhibitory effect of these phenolic acids occurs on the ATP-generating pathway. Caffeic and chlorogenic acids partially uncoupled oxidative phosphorylation in isolated mitochondria from *Solanum tuberosum* tubers (Makovec and Sindelár 1984). Also, cinnamic, caffeic and vanillic acids inhibited succinate dehydrogenase; complex II of the mitochondrial respiratory chain. On the other hand, gallic acid increased the activity of malate dehydrogenase, succinate dehydrogenase and glutamate dehydrogenase, but decreased cytochrome *c* oxidase activity.

Ferulic acid inhibited L-malate oxidation in *G. max* isolated mitochondria in acidic (pH 6.8) and alkaline (pH 7.8) conditions (Sert et al. 1997). Independently of the pH, both basal respiration (absence of ADP) and coupled respiration (presence of ADP) decreased with increasing concentrations of ferulic acid. However, maximal inhibition of rates of oxygen uptake (approximately 50%) were achieved at 5.0 mM ferulic acid. Therefore, inhibition of L-malate oxidation in mitochondria is unlikely to be one of the mechanisms of action of ferulic acid in plants. Interestingly, in

acidic conditions, the oxidation of L-malate occurred mainly through the NAD^+-linked malic enzyme, whereas malate dehydrogenase activity predominated in alkaline conditions. Furthermore, ferulic acid inhibited the production of pyruvate or oxaloacetate at both acidic and alkaline pH. In disagreement with Sert et al. (1997), 0.1 to 10 mM of ferulic, *p*-coumaric and vanillic acids did not affect the respiration of mitochondria isolated from *G. max* hypocotyls (Abrahim et al. 2003).

In experiments conducted on light-induced electron transport and ATP synthesis, benzoic and cinnamic acids derivatives slightly inhibited the CO_2-dependent O_2 evolution of intact chloroplasts of *S. oleracea* (Moreland and Novitzky 1987). Further experiments revealed that phenolic acids can inhibit coupled electron transport and phosphorylation. Notably, benzoic acid derivatives were weaker inhibitors than cinnamic acid derivatives. However, these findings did not allow the direct effects of phenolic acids on the electron transport chain or their indirect effects on ATP phosphorylation to be identified. For this, subsequent assays were made with uncouplers (NH_4Cl and carbonyl cyanide *p*-trifluoromethoxyphenylhydrazone; FCCP), which dissipate the transmembrane proton gradient. Under these conditions, the inhibition of electron transport coupled with ATP phosphorylation should be circumvented by uncouplers if phenolic acids act indirectly on the phosphorylation pathway. On the other hand, if phenolics directly affect the electron transport pathway, the uncouplers would not circumvent the inhibition of electron transport coupled with ATP phosphorylation. The results revealed that uncoupled electron transport was only partially relieved. In brief, this suggests that phenolic acids exert an inhibitory effect mainly on the ATP-generating pathway and a weaker effect on the electron transport pathway. Unfortunately, these results could not identify the specific site of action of phenolic acids.

Lignin metabolism

Exogenously applied hydroxycinnamic acids can inhibit plant growth primarily by altering lignin metabolism via two different mechanisms: (1) incorporation into lignin through the phenylpropanoid pathway, followed by premature cell wall lignification and/or (2) direct copolymerization with the canonical monolignols of the lignin structure.

The first study suggesting that a reduction of root growth by hydroxycinnamic acids is associated with premature cell wall lignification was described by Devi and Prasad (1996). Following this premise, Ferrarese et al. (2000) demonstrated that ferulic acid is rapidly absorbed by *G. max* roots. Hamada et al. (2003) found that ferulic acid, exogenously supplied, is converted to coniferyl and sinapyl alcohols in *Populus alba* callus. Also, Yamauchi and Fukushima (2004) revealed that ferulic acid labelled radioactively was incorporated in the G and S fractions of lignin in *Robinia pseudoacacia*.

Using *G. max* seedlings as a model, our group confirmed that the short-term treatment (24–72 h) with ferulic acid increases lignin contents with a consequent reduction of root growth and biomass (dos Santos et al. 2004). In addition, ferulic acid induced the early lignin production, affecting cell growth. To strengthen these findings, *G. max* seedlings were exposed to ferulic acid and specific enzyme inhibitors of the phenylpropanoid pathway and the lignin monomeric composition were determined in roots (dos Santos 2008; dos Santos et al. 2008a, b). These experiments clearly demonstrated that ferulic acid, when applied exogenously, stimulated the phenylpropanoid pathway followed by an uncontrolled lignification of vessels and fibers. In this case, the enzyme 4CL (Fig. 4) can be considered the gateway for ferulic acid, which is channeled as feruloyl-CoA in the metabolic pathway, inducing the biosynthesis of monolignols. A careful analysis of esterified phenolic acids revealed that in addition to inducing lignin formation (mainly G monomer), ferulic acid is incorporated directly into the cell wall and is probably esterified to the polysaccharides.

Applying the same strategy described for ferulic acid, the effects of other hydroxycinnamic acids on lignin monomeric composition were evaluated in *G. max* roots (Lima et al. 2013). The data revealed that cinnamic acid, the immediate precursor of hydroxycinnamic acids, is incorporated into the phenylpropanoid pathway. In this case, the channeling of the allelochemical in the metabolic pathway occurs by the enzyme C4H (Fig. 4). In fact, while ferulic acid raised the G monomer content, as previously described, cinnamic acid increased the H monomer content (Fig. 4). Similar effects were noted in seedlings treated with *p*-coumaric acid, i.e., significant increases of monomers H and G. However, seedlings treated

with *p*-coumaric acid plus methylenedioxycinnamic acid (MDCA), a 4CL enzyme inhibitor, did not increased H and G. These results suggest that channeling of *p*-coumaric acid into the metabolic pathway occurs through the reaction of 4CL. Caffeic acid also increased the H and G monomers of lignin. The inhibition of 4CL by MDCA interrupted the entry and conversion of caffeic acid to G monomer, without affecting the production of H. These results suggest that caffeic acid can be channeled into the metabolic pathway by two ways: by a CoA-independent way leading to production of H and, by another, CoA-dependent, which channels it to the formation of G. Finally, the sinapic acid did not affect the growth of *G. max* roots or its lignin contents. However, this compound significantly increased G and S, which strengthens the hypothesis of channeling the hydroxycinnamic acids into the phenylpropanoid pathway. Briefly, all these findings reinforce the mechanism of action proposed by our research group. In this proposed model, hydroxycinnamic acids affect the *G. max* growth by means of the phenylpropanoid pathway, which leads to the lignin formation as its final product (Fig. 4). Among other effects, the uncontrolled incorporation of hydroxycinnamic acids into the phenylpropanoid pathway, with high lignin production in the cell wall, restricts cell expansion and plant growth.

As mentioned above, some hydroxycinnamic acids or their derivatives can be directly copolymerized with canonical monolignols into lignin structure. For instance, rosmarinic acid, an ester of caffeic acid with 3,4-dihydroxyphenyl-lactic acid, was copolymerized (peroxidase-catalyzed reaction) with monolignols in in vitro experiments (Tobimatsu et al. 2012). Similar copolymerization assays have been shown for methyl caffeate, and caffeoylquinic and feruloylquinic acids (Grabber et al. 2010). Notably, in an *in planta* study, short-term exposure (24 h) to rosmarinic acid reduced growth and altered the lignin monomer composition of *Zea mays* roots (Bevilaqua et al. 2019), suggesting that its copolymerization into lignin can also occur in vivo.

Secondary effects of phenolic acids on plants

Water relations

Exposure of plants to phenolic acids often induces deficiencies in water absorption. There is a consensus that water stress caused by phenolic acids is a result of the action of these compounds on the plasma membrane (Einhellig 2004). Among the main analytical tools used to evaluate the effects of phenolic acids on water absorption are the determination of the relative water content (RWC) and the water potential of the tissues.

The physiological process most affected by the reduction of water potential is cell expansion. It is very common for plants exposed to phenolic acids to show an impairment in water absorption and a significant reduction of growth. For example, the growth-inhibition of *Euphorbia esula* exposed to caffeic acid was attributed to modifications in the water relations of the plant (Barkosky et al. 2000). Cinnamic acid also reduced the leaf water potential in *Lolium perenne*, *Dactylis glomerata* and *Rumex acetosa*, which may have contributed to the reduction of growth (Hussain and Reigosa 2011).

Changes in water status are also felt at the stomatal level. One commonly used approach to assess stomatal resistance, which identifies possible water stress, is determination of the isotopic ratio of carbon. This method is based on fact that greater stomatal restrictions lead to lower discrimination against the heavier carbon isotope (^{13}C). *E. esula* plants exposed to caffeic acid presented greater diffusive resistance to CO_2 and lower transpiration rates than control plants (Barkosky et al. 2000). Although the exact mechanism of stomatal closure has not been determined, the authors suggest the involvement of the allelochemical on the cell membrane. As a probable cause, the induced changes in the water relations of *E. esula* may be due to direct action of caffeic acid on ions uptake or have been indirectly caused by mineral deficiency.

The *p*-hydroxybenzoic acid also induced water stress in *G. max* plants grown hydroponically (Barkosky and Einhellig 2003). The first indication of water stress was a reduction of stomatal conductance. The treated plants had lower leaf water potential at the end of the treatment (28 days of cultivation) and modifications in the isotopic ratio of carbon in the tissues due to the high stomatal resistance, i.e., evidences of water stress. Ferulic, *p*-coumaric, caffeic, salicylic and gallic acids caused similar results in *G. max* (Einhellig 2004). On the other hand, ferulic and *p*-hydroxybenzoic acids do not alter the relative water content and osmotic potential of *Lactuca sativa* leaves (Hussain et al. 2010).

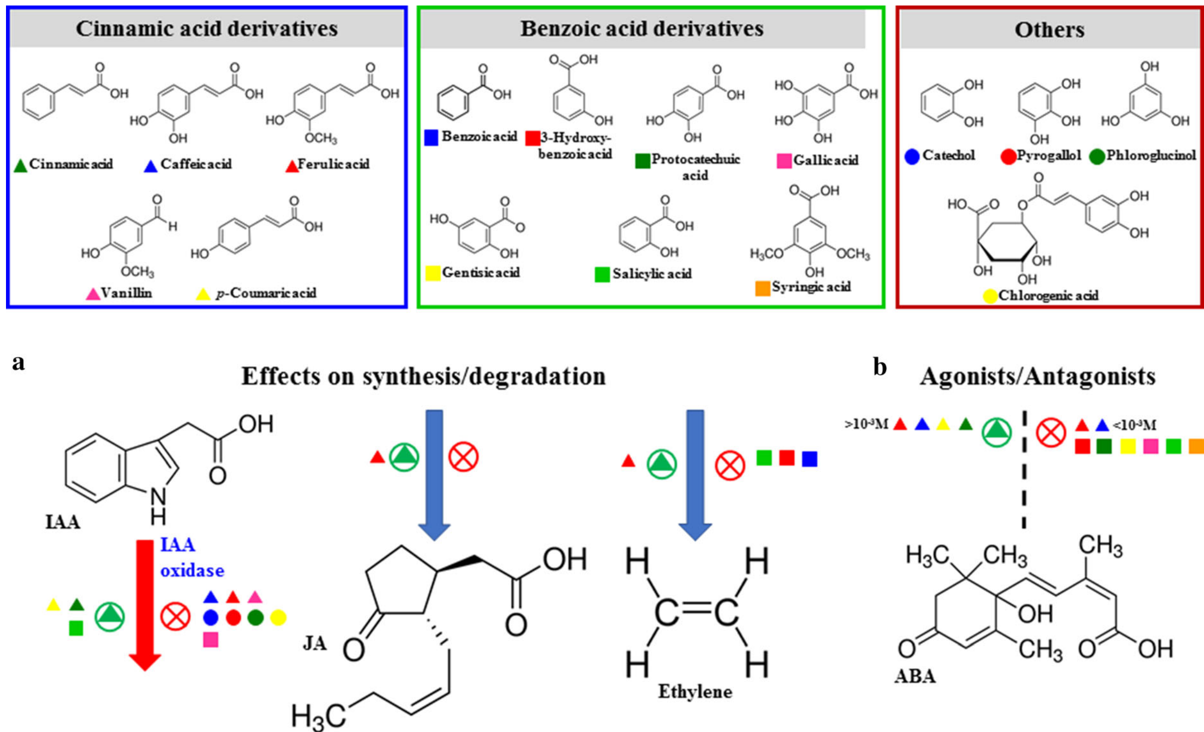


Fig. 7 Effects of simple phenolic acid on phytohormones. Effects of simple phenolic acid on biosynthesis (blue arrows) and degradation (red arrows) of indole acetic acid (IAA), jasmonic acid (JA), and ethylene (**a**). For IAA, only the decarboxylation pathway of degradation, catalyzed by IAA oxidase, is shown. Simple phenolic that act as agonists/antagonists of abscisic acid (**b**). Cinnamic and benzoic acid derivatives and other phenolics are represented by colored symbols i.e., triangles, squares, and circles, respectively. In (**a**) and (**b**), symbols are used to represent the respective simple phenolic acids. The green () and red () symbols added parallel to the arrows refer to stimulatory/agonist or inhibitory/antagonist effects, respectively

Hormonal balance

In some situations, the exposure of plants to phenolic compounds induces morphological changes that reflect variations in the hormonal balance (Fig. 7). Several studies suggest that phenolic compounds may induce the rooting. Since auxins are the major hormones required for this process, it is assumed that phenolic compounds induce root formation by influencing the metabolism of indole-3-acetic acid (IAA). This hormone can be inactivated by conjugation or decarboxylation; the latter catalyzed by the enzyme IAA oxidase. In some cases, phenolic compounds can stimulate rooting by preventing decarboxylation of IAA. In others, they can act as cofactors of the IAA oxidase stimulating their degradation (Fig. 7a).

The effect probably exerted by a phenolic compound on metabolism of IAA is revealed by structural analysis. Some authors suggest that the decarboxylation of IAA is inhibited by *meta*-, *ortho*-diphenols and polyphenols (Wilson and Van Staden 1990; Bandurski et al. 1995). On the other hand, monophenols appear to stimulate the degradation of IAA by decarboxylation. De Klerk et al. (2011) reported that phenolic compounds increase the rooting in stem slices of *Malus* 'Jork 9′, particularly in the presence of sub-optimal concentrations of IAA. The effect was observed for *o*-diphenols (caffeic and chlorogenic acids, and catechol), methylated *o*-diphenols (ferulic acid and vanillin) and triphenols (gallic and tannic acids, phloroglucinol and pyrogallol). The induction of rooting seems to be related to the massive inhibition of decarboxylation of IAA. The main evidences that support this hypothesis are: (1) the stimulation of rooting in the presence of IAA, but not of α-naphthalene acetic acid (NAA), which is an auxin that, unlike IAA, is inactivated only by conjugation and not by decarboxylation; (2) a stronger rooting with IAA, which does not occur with NAA; (3) the optimum concentration of IAA that induced rooting

was lower in the presence of phenolic acids. In addition, *p*-coumaric acid had no effect, but salicylic acid increased the decarboxylation of IAA. Furthermore, there is evidence that monophenols act as cofactors for IAA oxidase, increasing the decarboxylation of IAA (Bandurski et al. 1995).

In a study conducted by our research group, it was noted that cinnamic acid significantly increased the activity of IAA oxidase in *G. max* roots (Salvador et al. 2013). The hypothesis is that this enzyme would contribute to the reduction of the endogenous levels of IAA, reducing the growth of *G. max* seedlings. In a previous work, it was shown that cinnamic acid antagonized the growth-promoting effect of IAA (Overbeek et al. 1951) or altered its biosynthesis (Einhellig 2004).

In contrast to the data found by de Klerk et al. (2011), ferulic acid increased the activity of IAA oxidase in *Z. mays* seedlings (Devi and Prasad 1996). The authors state that peroxidases, which also had their activities stimulated by ferulic acid treatment, may have contributed to the degradation of endogenous IAA. In fact, this is a well-established function of peroxidases (Gebhardt 1982; Zheng and Van Huystee 1992).

A microarray analysis revealed that ferulic acid causes overexpression of 972 genes in *O. sativa* roots (Chi et al. 2013). Among the activated genes, identified by GO analysis, were those involved in the biosynthesis of jasmonic acid and ethylene (Fig. 7a). The treatment of *O. sativa* plants with ferulic acid also inhibited the formation of lateral roots and root hairs and suppressed the expression of two related genes. According to the RiceXPro database, it is possible that the jasmonic acid has inhibited the expression of these genes and, therefore, caused morphological changes that followed the exposure of *O. sativa* roots to ferulic acid (Chi et al. 2013). In fact, the treatment of *A. thaliana* with jasmonic acid and ethylene inhibited the root elongation (Staswick et al. 1992; Hua and Meyerowitz 1998).

Ethylene production from 1-aminocyclopropane-1-carboxylate (ACC) was strongly inhibited by SA in a *Pyrus communis* cell culture (Leslie and Romani 1988). The authors stated that *o*-hydroxylation should be an important characteristic for the activity of salicylic acid since benzoic acid derivatives monosubstituted at position 3 or 4 only inhibited ethylene biosynthesis at high concentrations. In contrast, 25 μM SA inhibited the production of ethylene.

In another study, SA derivatives (salicyl alcohol and salicin), whose carboxyl is not present, have failed to inhibit ethylene production. These data suggest that this functional group should play an important role in determining the activity of these compounds. The *p*-amino salicylate and 3,5-dinitrosalicylate derivatives also showed lower activities than SA. It is possible that the presence of other functional groups may interfere in the binding of the molecule to its molecular target. An inhibitory effect of benzoic acid on ethylene production was observed in green *S. lycopersicum* and *Malus domestica* (Baker et al. 1978).

Phenolic acids can, antagonistically, influence the effects of abscisic acid (ABA) (Fig. 7b). This hormone plays important regulatory roles in the beginning and maintenance of dormancy, stress responses, vegetative growth, senescence and stomatal control. Like IAA, the structure of phenolic acid is directly related to its effects on ABA activity. In this context, valuable information was obtained from the work of Li et al. (1993), in which mono- or dihydroxylated phenolic acids (cinnamic, *p*-coumaric, caffeic and ferulic acids at $> 10^{-3}$ M), applied together with ABA, caused additive inhibitory effects on the growth of *L. sativa* seedlings. In contrast, caffeic and ferulic acids (at $< 10^{-3}$ M) reversed the growth-inhibition caused by low concentrations of ABA (10^{-5} M), but not high concentrations. Although the mechanism involved is unclear, monohydroxylated phenolic acids inhibit, while di- or polyhydroxylates (at low concentrations) promote the growth of *L. sativa* seedlings influencing the action of ABA.

The ABA causes stomatal closure due to inhibition of K^+ uptake and increase of ion efflux from guard cells. Various hydroxybenzoic and hydroxycinnamic acids were able to reverse the effects of ABA (Purohit et al. 1991). All hydroxybenzoic derivatives (3-hydroxybenzoic, protocatechuic, syringic, salicylic, gentisic, gallic and tannic acids) restored the stomatal closure induced by 10 μM ABA; vanillic acid was the most effective. The presence of hydroxyls or methoxyls at different positions of the aromatic ring of benzoic acid influenced the effect of ABA. For example, 3-hydroxybenzoic acid did not substantially reverse the effects of ABA. On the other hand, protocatechuic (with an additional hydroxyl in C4) and vanillic (with a C3 methoxyl and a C4 hydroxyl)

acids inhibited ABA. In addition, the syringic acid (with an additional methoxyl group at C5) showed little effect. Similarly, hydroxycinnamic derivatives showed greater effects due to the presence of a hydroxyl in the *para* position; ferulic and caffeic acids were more effective against ABA than cinnamic acid. In contrast to hydroxybenzoic derivatives, C3 and C5 methoxylation did not modulate the effects of hydroxycinnamic derivatives. In addition, hydroxycinnamic acids (hydrocinnamic and hydrocaffeic acids) did not reverse the effects of ABA, suggesting that double bonding in the side chain of cinnamic acid is essential in this case (Purohit et al. 1991).

Oxidative stress and related enzymes

The ROS are constantly produced in all cell compartments during aerobic metabolism. However, the PSI and PSII reaction centers in chloroplasts are the major generation site of ROS (Asada 2006). The superoxide anion ($O_2^{\cdot-}$) and hydrogen peroxide (H_2O_2) are produced by the photoreduction of oxygen in PSI, while the excited singlet oxygen ($^1O_2$) evolves from PSII. The ROS also include the hydroxyl ($^{\cdot}OH$) and perhydroxyl ($^{\cdot}O_2H$) radicals. When plants are exposed to stress, the production of ROS increases due to an imbalance of cellular homeostasis (Sharma et al. 2012). Small changes in ROS levels lead to changes in cellular metabolism in order to maintain redox homeostasis. When the production of ROS exceeds the antioxidant capacity, the cells undergo oxidative damage. To avoid this, plants have developed enzymatic and non-enzymatic defense mechanisms, in which metabolites and enzymes with antioxidant roles act to neutralize ROS (Shin et al. 2013).

Simple phenolic acids can affect the redox *status* of cells, acting as antioxidants or inducing oxidative stress (Dai et al. 2012; Rajput and Rao 2013). Factors that determine one or another action include the concentration and type of phenolic acid, the exposure time and the plant species.

The effect of vanillic acid on the SOD, POD, CAT and polyphenol oxidase (PPO) activities was dose-dependent (Ghareib et al. 2010). Low concentrations of vanillic acid stimulated the enzyme activities and reduced the contents of phenolic compounds, H_2O_2 and lipid peroxidation; the opposite occurred at higher concentrations. POD activity was also dose-dependently stimulated in *O. sativa* plants exposed to vanillic acid (Jazayeri et al. 2007).

The effects of phenolic acids on the antioxidant system of plants can confer or increase the tolerance in stress situations. For example, caffeic acid pretreatment increased the tolerance of *C. sativus* seedlings to dehydration (Wan et al. 2014). The protective effect seems to be related to the reduction of $O_2^{\cdot-}$, H_2O_2 and malondialdehyde (MDA) contents in seedlings pretreated with the phenolic acid. These effects probably resulted from increased activities of SOD, POD, glutathione peroxidase (GPX), ascorbate peroxidase (APX), glutathione reductase (GR), dehydroascorbate reductase (DAR) and monodehydroascorbate reductase (MDAR) in plants pretreated with caffeic acid. In another study, the treatment of *O. sativa* plants with gallic acid minimized the damage caused by salt (NaCl) and/or osmotic (polyethylene glycol) stresses. The two stressing conditions increased H_2O_2 production and lipid peroxidation. Under salt stress, the levels of lipid peroxidation were reduced by treatment with gallic acid. Increases in the activities of SOD, POD and APX enzymes were important to avoid oxidative damage; an indicator that gallic acid stimulated responses of the cellular antioxidant system.

The pretreatment of *C. sativus* plants with 50 μM cinnamic acid also increased the ability of plants to resist water stress induced by polyethylene glycol (PEG). Water stress increased the H_2O_2 and MDA contents in the control plants and reduced in plants pretreated with cinnamic acid. It is possible that the mechanism involved in the acquisition of stress tolerance is related to the activation of the antioxidant system and accumulation of free sugars in the leaves. In fact, the authors observed increases in the activities of SOD, CAT, guaiacol peroxidase (GP), GR, GPX, and DHAR (Sun et al. 2012). In addition, a possible action of sugars as ROS-scavengers has been suggested and described in the literature (Couée et al. 2006; Bolouri-Moghaddam et al. 2010).

Activations of SOD, CAT, GP, and GR were observed in *Vigna radiata* exposed to caffeic acid. Caffeic acid also increased H_2O_2 and MDA contents (Singh et al. 2009). Cinnamic acid reduced oxidative damage in *Z. mays* plants exposed to NaCl, probably through increased SOD activity (Singh et al. 2013). In contrast, the activity of CAT and the content of MDA were significantly reduced in leaves. In *G. max*, cinnamic acid decreased the H_2O_2 content in roots

(Bubna et al. 2011). However, soluble and cell wall-bound POD activities, as well as the lignin content, were enhanced by treatment with the allelochemical.

It is a fact that a phenolic acid can be phytotoxic to an only plant species without affecting others. For example, cinnamic acid activated SOD, CAT, POD and NADPH oxidase and increased the production of $O_2^{\cdot -}$ and H_2O_2, inducing the cell death in *C. sativus* roots, but without any effect on *Cucurbita ficifolia* (Ding et al. 2007a, b).

In *O. sativa* roots, ferulic acid increased the levels of ROS, lipid peroxidation and the activities of three isoenzymes of lipoxygenase, as well as intracellular calcium levels. Calcium acts in an integrated manner with ROS and, therefore, it functions as a secondary messenger in the activation of defense genes in response to the allelochemical (Chi et al. 2013).

Photosynthesis

One of the main effects caused by phenolic acids on photosynthesis is the reduction of chlorophyll content (Li et al. 2010). Several hydroxycinnamic and hydroxybenzoic acids, such as cinnamic, *p*-coumaric, caffeic, ferulic, benzoic, *p*-hydroxybenzoic, protocatechuic and syringic acids, reduced chlorophyll content in *G. max*, *Vigna sinensis* (Alsaadawi et al. 1986), *O. sativa* (Yang et al. 2002, 2004) and *Echinochloa crus-galli* (Esmaeili et al. 2012).

Changes in chlorophyll content can be due to a reduction of its biosynthesis and/or increase of its degradation. Yang et al. (2012) suggested that *o*-hydroxyphenylacetic, *p*-coumaric and ferulic acids reduce chlorophyll content in *O. sativa* due to inhibition of Mg-chelatase, which converts protoporphyrin IX to Mg-protoporphyrin IX in the chlorophyll biosynthetic pathway. In an earlier study, these authors noted that the same phenolic compounds stimulated the activity of chlorophyllase and of Mg-dechelatase, which are active in the degradation of chlorophyll (Yang et al. 2004). However, it has not been confirmed whether the inhibition of Mg-chelatase and stimulation of chlorophyllase and Mg-dechelatase are primary effects of phenolic acids or only consequences of other biological events.

Several studies have shown that phenolic acids are capable of inhibiting or stimulating the photosynthetic rate. This effect is due to changes in stomatal conductance, although the chlorophyll content may contribute in some cases (Einhellig 2004). Hydroxycinnamic and hydroxybenzoic acids significantly reduced the photosynthetic rate, stomatal conductance, transpiration and intercellular CO_2 concentration in *C. sativus* seedlings (Yu et al. 2003). Reductions ranged from 7 to 50% for photosynthetic rate and 51% to 87% for transpiration. Among the analyzed parameters, the stomatal conductance was the most affected, which indicates to be the factor responsible for the reductions of the photosynthetic rate and transpiration. For these authors, it is probable that allelochemicals do not act directly on photosynthesis but induce the peroxidation of membrane lipids. In fact, the activities of SOD and POD enzymes and MDA contents increased in plants exposed to the allelochemicals. These data reinforce the hypothesis that these allelochemicals induce oxidative stress, which, consequently, limit photosynthesis.

Stimulatory effects on photosynthetic rate were observed after foliar application of salicylic, acetylsalicylic and gentisic acids in *G. max* and *Z. mays* (Khan et al. 2003). The photosynthetic rate, stomatal conductance and transpiration increased without any change in chlorophyll content. Leaf area and dry biomass increased, although the height and length of the roots were not affected. The initial stimulus in the photosynthetic rate was accompanied by reduction of the intercellular CO_2 concentration in treated plants. This suggests the greater use of CO_2 by chloroplasts (possible stimulus in carbon assimilation reactions) and reveals that the stimulus of photosynthetic rate cannot be due to the stomatal opening alone. In addition, it is possible that increased stomatal conductance is related to the ability of salicylic acid to reverse the effects of ABA. A similar trend has been noted in *Z. mays* plants after combined stem-injection of salicylic acid and sucrose (Zhou et al. 1999).

In some situations, phenolic acids induce biochemical changes that make the plants more resistant to environmental stresses. For example, less drastic effects on photosynthesis, chlorophyll *a* content and growth were noted in *G. max* plants under drought and exposed to benzoic acid (Anjum et al. 2013). The increase in the photosynthetic rate was associated with enhanced stomatal conductance, which may be a direct cause of the higher photosynthetic activity. Treatment with benzoic acid contributed to the survival of plants exposed to drought, although it did not eliminate stress. Partially, the maintenance of the

high photosynthetic activity was related to the protection of the photosynthetic apparatus against oxidative damage. In addition, cinnamic acid activated some enzymes that scavenge ROS. In agreement with all these findings, salicylic acid increased photosynthetic rate, stomatal conductance and transpiration in *Brassica juncea* (Fariduddin et al. 2003). Additionally, salicylic acid induced tolerance to salinity in *S. lycopersicum* (Stevens et al. 2006). The protective effect against NaCl stress was related to increased photosynthetic rate, stomatal conductance and transpiration, as well as protection of membrane integrity (Stevens et al. 2006).

Effects of phenolic acids on chlorophyll *a* fluorescence have been described. In leaves of *L. perenne*, *D. glomerata* and *R. acetosa*, cinnamic acid reduced the maximum quantum yield of PSII (F_v/F_m) and the quantum yield of PSII under light (Φ_{PSII}). The allelochemical reduced the relative water content (RWC) of the leaves inducing the stomatal closure. This, in turn, reduced the CO_2 supply to mesophyll cells and, consequently, photosynthesis. The lower availability of CO_2 inhibited the rates of consumption of ATP and NADPH by the reactions of carbon assimilation and, therefore, the rate of electron transport, culminating in a reduction of Φ_{PSII}. Similarly, reductions in photosynthetic rate and Rubisco activity (without any effect on chlorophyll fluorescence and chlorophyll content) were observed in *Vigna unguiculata* plants exposed to cinnamic acid (Huang and Bie 2010). In addition, cinnamic acid decreases the photosynthetic rate, stomatal conductance and Φ_{PSII} in *C. sativus* (Ye et al. 2004). A similar trend has been observed in *L. sativa* exposed to *p*-hydroxybenzoic and ferulic acids (Hussain et al. 2010).

More recently, Lu et al. (2018) evaluated the effects of the exogenous application of phthalic and syringic acids (1 to 27 μg g^{-1} of soil) on the photosynthetic capacity of strawberry (*Fragaria* × *ananassa* Duch., cultivar Hokowase) plants. At low concentration, phthalic acid stimulated the biosynthesis of photosynthetic pigments (chlorophylls and carotenoids) and stomatal conductance and slightly increased the photosynthetic rate. The inverse effect, however, was noted at higher concentration, i.e., there was a decrease in the biosynthesis of chlorophyll, photosynthetic rate, stomatal conductance and internal CO_2 concentration. Similarly, syringic acid inhibited the biosynthesis of pigments and affected gas exchange parameters. At high concentrations, both phenolic acids reduced the photochemical quenching coefficient and, therefore, primary light energy conversion by PSII and the electron transport rate. Consequently, ATP synthesis, the carbon assimilation rate and accumulation of biomass in strawberry were impaired. In brief, these findings suggest that syringic and phthalic acids reduce the photosynthetic rate, mainly due to stomatal limitation and lowered pigment biosynthesis but with significant inhibition of carbon assimilation reactions at higher concentrations.

Cellular respiration

At the seedling stage and in heterotrophic tissues, essentially all metabolic energy needed for cellular activities comes from cellular respiration. Therefore, inhibition of mitochondrial respiration activity may affect several ATP-dependent processes, which are essential for plant growth and development (Ishii-Iwamoto et al. 2006). As previously described, phenolic acids can directly act as inhibitors or weak uncouplers of oxidative phosphorylation, and enhanced production of ROS is the main secondary effect. In fact, $O_2^{\cdot-}$ generation in complex I and III of the respiratory chain was increased by biotic and abiotic stresses and by inhibition of cytochrome and alternative oxidases (Popov et al. 1997; Parsons et al. 1999; Mittova et al. 2003). Increased ROS generation was noted in the roots of *A. thaliana* exposed to rosmarinic acid, a derivative of caffeic acid (Araniti et al. 2018). Rosmarinic acid also reduced CAT and SOD activities, produced ROS, increased MDA formation and dissipated the mitochondrial transmembrane potential ($\Delta\Psi_m$). A metabolomic approach revealed a significant decrease in some metabolites of the citric acid cycle, and of amino acids biosynthesized from them. In brief, all these effects profoundly altered the mitochondrial energy metabolism and inhibited the growth and development of roots and their morphology. In support, the exogenous application of syringic and phthalic acids increased the production of H_2O_2 and $O_2^{\cdot-}$ in strawberry (*Fragaria* × *ananassa* Duch., cultivar Hokowase) roots, while the activities of POD and SOD were markedly reduced (Wang et al. 2019). At mitochondrial level, an increase in the degree of opening of the mitochondrial membrane permeability transition pore (MPTP) and

the decrease in $\Delta\Psi_m$ and cytochrome c/a ratio were evident. These effects strongly suggest the loss of integrity of the mitochondrial membrane induced by ROS. As previously discussed, phenolic acids, directly or not, oxidize sulfhydryl groups in integral membrane proteins by inducing the formation of disulfide bonds (Baziramakenga et al. 1995). It is known that ROS promotes the opening of MPTPs through the oxidation of sulfhydryl groups in MPTP-related proteins (Halestrap 2009). Thus, syringic and phthalic acids profoundly affect mitochondrial activity, and the energy conversion processes, by compromising the integrity of its membranes (Wang et al. 2019).

In *G. max*, cinnamic acid reduced the rate of oxygen consumption in cotyledons (Peñuelas et al. 1996). On the other hand, Dedonder and Van Sumere (1971) had earlier noted that cinnamic acid stimulated the respiration of *Chlorella vulgaris* (green algae). The application of salicylic acid reduced the respiratory activity in *Beta vulgaris* minimally processed in relation to those not processed (Kluge et al. 2010). According to the authors, salicylic acid possibly reduces the cytosolic pH and slows glycolytic velocity.

In summary, phenolic acids appear to affect cellular respiration in the following way: (1) their inhibitory or uncoupling activity on electron transport leads to the enhanced production of ROS; (2) the ROS produced oxidizes the sulfhydryl groups of important membrane proteins with subsequent changes in membrane permeability; (3) ROS-induced lipid peroxidation affects membrane integrity and dissipates $\Delta\Psi_m$, and (4) notable changes in the activity of the enzymes of the acid citric cycle and related metabolites impair mitochondrial energy metabolism.

Conclusions

Herein, we have reviewed the main metabolic pathways that lead to the production of simple phenolic acids in plants, among them the shikimate and phenylpropanoid pathways. In addition, the biosynthesis of benzoic acids, including vanillin and salicylic acid was detailed. The most relevant effects of simple phenolic acids on different plant species were described. High biodegradability and strong adsorption by soil particles seem to be the main factors that limit the allelopathic action of phenolic acids in the environment. Furthermore, most studies reported to date describe secondary effects from a primary event. Thus, further efforts to identify the primary molecular target of phenolic acids on plants are necessary to understand the ecological role of these compounds.

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References

Abrahim D, Takahashi L, Kelmer-Bracht AM, Ishii-Iwamoto EL (2003) Effects of phenolic acids and monoterpenes on the mitochondrial respiration of soybean hypocotyl axes. Allelopath J 11:21–30

Ahmed MA, El-Mawla A, Beerhues L (2002) Benzoic acid biosynthesis in cell cultures of *Hypericum androsaemum*. Planta 214:727–733. https://doi.org/10.1007/s004250100657

Alber A, Ehlting J (2012) Cytochrome P450s in lignin biosynthesis. In: Jouanin L, Lapierre C (eds) Lignins: biosynthesis, biodegradation and bioengineering. Elsevier, London, pp 113–143

Alsaadawi IS, Al-Hadithy SM, Arif MB (1986) Effects of three phenolic acids on chlorophyll content and ions uptake in cowpea seedlings. J Chem Ecol 12:221–227. https://doi.org/10.1007/BF01045605

Anjum SA, Ehsanullah XL et al (2013) Exogenous benzoic acid (BZA) treatment can induce drought tolerance in soybean plants by improving gas-exchange and chlorophyll contents. Aust J Crop Sci 7:555–560

Antoniw JF, White RF (1980) The effects of aspirin and polyacrylic acid on soluble leaf proteins and resistance to virus infection in five cultivars of tobacco. J Phytopathol 98:331–341. https://doi.org/10.1111/j.1439-0434.1980.tb03748.x

Araniti F, Costas-Gil A, Cabeiras-Freijanes L et al (2018) Rosmarinic acid induces programmed cell death in Arabidopsis seedlings through reactive oxygen species and mitochondrial dysfunction. PLoS ONE 13:1–26. https://doi.org/10.1371/journal.pone.0208802

Asada K (2006) Production and scavenging of reactive oxygen species in chloroplasts and their functions. Plant Physiol 141:391–396. https://doi.org/10.1104/pp.106.082040

Bahrami SN, Zakizadeh H, Hamidoghli Y, Ghasemnezhad M (2013) Salicylic acid retards petal senescence in cut lisianthus (*Eustoma grandiflorum* 'Miarichi Grand White') flowers. Hortic Environ Biotechnol 54:519–523. https://doi.org/10.1007/s13580-013-1058-y

Baker JE, Lieberman M, Anderson JD (1978) Inhibition of ethylene production in fruit slices by a rhizobitoxine analog and free radical scavengers. Plant Physiol 61:886–888. https://doi.org/10.1104/pp.61.6.886

Bandurski RS, Cohen JD, Slovin J, Reinecke DM (1995) Auxin biosynthesis and metabolism. In: Davies PJ (ed) Plant hormones—physiology, biochemistry and molecular biology. Springer, New York, pp 39–65

Barkosky RR, Einhellig FA (2003) Allelopathic interference of plant-water relationships by *p*-hydroxybenzoic acid. Bot Bull Acad Sin 44:53–58

Barkosky RR, Einhellig FA, Butler JL (2000) Caffeic acid-induced changes in plant-water relationships and photosynthesis in leafy spurge *Euphorbia esula*. J Chem Ecol 26:2095–2109. https://doi.org/10.1023/A:1005564315131

Barros J, Escamilla-Trevino L, Song L et al (2019) 4-Coumarate 3-hydroxylase in the lignin biosynthesis pathway is a cytosolic ascorbate peroxidase. Nat Commun 10:1–11. https://doi.org/10.1038/s41467-019-10082-7

Barros J, Serrani-Yarce JC, Chen F et al (2016) Role of bifunctional ammonia-lyase in grass cell wall biosynthesis. Nat Plants 2:1–9. https://doi.org/10.1038/NPLANTS.2016.50

Bartsch M, Bednarek P, Vivancos PD et al (2010) Accumulation of isochorismate-derived 2,3-dihydroxybenzoic 3-*O*-β-D-xyloside in *Arabidopsis* resistance to pathogens and ageing of leaves. J Biol Chem 285:25654–25665. https://doi.org/10.1074/jbc.M109.092569

Baziramakenga R, Leroux GD, Simard RR (1995) Effects of benzoic and cinnamic acids on membrane permeability of soybean roots. J Chem Ecol 21:1271–1285. https://doi.org/10.1007/BF02027561

Bellés JM, Garro R, Fayos J et al (1999) Gentisic acid as a pathogen-inducible signal, additional to salicylic acid for activation of plant defenses in tomato. Mol Plant Microbe Interact 12:227–235. https://doi.org/10.1094/MPMI.1999.12.3.227

Bevilaqua JM, Finger-Teixeira A, Marchiosi R et al (2019) Exogenous application of rosmarinic acid improves saccharification without affecting growth and lignification of maize. Plant Physiol Biochem 142:275–282. https://doi.org/10.1016/j.plaphy.2019.07.015

Bhattacharya S, Kumar P (2012) An in silico approach to structural elucidation of 3-deoxy-D-arabino-heptulosonate 7-phosphate synthase from *Arabidopsis thaliana*: hints for herbicide design. Phytochemistry 73:7–14. https://doi.org/10.1016/j.phytochem.2011.09.005

Bischoff M, Schaller A, Bieri F et al (2001) Molecular characterization of tomato 3-dehydroquinate dehydratase-shikimate: NADP oxidoreductase. Plant Physiol 125:1891–1900. https://doi.org/10.1104/pp.125.4.1891

Blum U (2004) Fate of phenolic allelochemicals in soils—the role of soil and rhizosphere microorganisms. In: Macías FA, Galindo JCG, Molinillo JMG, Cutler HG (eds) Allelopathy: Chemistry and mode of action of allelochemicals. CRC Press, Boca Raton

Boerjan W, Ralph J, Baucher M (2003) Lignin biosynthesis. Annu Rev Plant Biol 54:519–546. https://doi.org/10.1146/annurev.arplant.54.031902.134938

Bolouri-Moghaddam MR, Le Roy K, Xiang L et al (2010) Sugar signaling and antioxidant network connections in plant cells. FEBS J 277:2022–2037. https://doi.org/10.1111/j.1742-4658.2010.07633.x

Bubna GA, Lima RB, Zanardo DYL et al (2011) Exogenous caffeic acid inhibits the growth and enhances the lignification of the roots of soybean (*Glycine max*). J Plant Physiol 168:1627–1633. https://doi.org/10.1016/j.jplph.2011.03.005

Budi Muljono RA, Scheffer JJC, Verpoorte R (2002) Isochorismate is an intermediate in 2,3-dihydroxybenzoic acid biosynthesis in *Catharanthus roseus* cell cultures. Plant Physiol Biochem 40:231–234. https://doi.org/10.1016/S0981-9428(02)01369-4

Castañeda-Ovando A, Pacheco-Hernández MDL, Páez-Hernández ME et al (2009) Chemical studies of anthocyanins: a review. Food Chem 113:859–871. https://doi.org/10.1016/j.foodchem.2008.09.001

Catinot J, Buchala A, Abou-Mansour E, Métraux JP (2008) Salicylic acid production in response to biotic and abiotic stress depends on isochorismate in *Nicotiana benthamiana*. FEBS Lett 582:473–478. https://doi.org/10.1016/j.febslet.2007.12.039

Cesarino I, Simões MS, dos Santos BM et al (2016) Building the wall: recent advances in understanding lignin metabolism in grasses. Acta Physiol Plant 38:269. https://doi.org/10.1007/s11738-016-2293-5

Chandran D, Rickert J, Huang Y et al (2014) Atypical E2F transcriptional repressor DEL1 acts at the intersection of plant growth and immunity by controlling the hormone salicylic acid. Cell Host Microbe 15:506–513. https://doi.org/10.1016/j.chom.2014.03.007

Chandrasekara A (2019) Phenolic acids. Encycl Food Chem. https://doi.org/10.1016/B978-0-08-100596-5.22395-0

Chee MJY, Lycett GW, Khoo TJ, Chin CF (2017) Bioengineering of the plant culture of *Capsicum frutescens* with vanillin synthase gene for the production of vanillin. Mol Biotechnol 59:1–8. https://doi.org/10.1007/s12033-016-9986-2

Chen Z, Zheng Z, Huang J et al (2009) Biosynthesis of salicylic acid in plants. Plant Signal Behav 4:493–496. https://doi.org/10.4161/psb.4.6.8392

Chi WC, Chen YA, Hsiung YC et al (2013) Autotoxicity mechanism of *Oryza sativa*: Transcriptome response in rice roots exposed to ferulic acid. BMC Genomics 14:351. https://doi.org/10.1186/1471-2164-14-351

Chipley JR (2005) Sodium benzoate and benzoic acid. In: Davidson PM, Sofos JN, Branen AL (eds) Antimicrobials in food, 3rd edn. Taylor & Francis, Boca Raton, pp 11–48

Chong J, Pierrel MA, Atanassova R et al (2001) Free and conjugated benzoic acid in tobacco plants and cell cultures. Induced accumulation upon elicitation of defense responses and role as salicylic acid precursors. Plant Physiol 125:318–328. https://doi.org/10.1104/pp.125.1.318

Coggins JR, Abell C, Evans LB et al (2003) Experiences with the shikimate-pathway enzymes as targets for rational drug design. Biochem Soc Trans 31:548–552. https://doi.org/10.1042/bst0310548

Coracini JD, de Azevedo WF (2014) Shikimate kinase, a protein target for drug design. Curr Med Chem 21:592–604. https://doi.org/10.2174/09298673113206660299

Couée I, Sulmon C, Gouesbet G, El Amrani A (2006) Involvement of soluble sugars in reactive oxygen species balance and responses to oxidative stress in plants. J Exp Bot 57:449–459. https://doi.org/10.1093/jxb/erj027

Dai AH, Nie YX, Yu B et al (2012) Cinnamic acid pretreatment enhances heat tolerance of cucumber leaves through modulating antioxidant enzyme activity. Environ Exp Bot 79:1–10. https://doi.org/10.1016/j.envexpbot.2012.01.003

Dalton BR (1999) The occurrence and behavior of plant phenolic acids in soil environmental and their potential involvement in allelochemical interference interaction: methodological limitations in establishing conclusive proof of allelopathy. In: Inderjit M, Dakshini KMM, Foy CL (eds) Principles and practices in plant ecology: allelochemical interactions. CRC Press, Boca Raton, pp 54–57

De Boer JG, Dicke M (2004) Prey searching behavior of the predatory mite *Phytoseiulus persimilis*: the role of methyl salicylate. J Chem Ecol 30(11):255–271. https://doi.org/10.1023/b:joec.0000017976.60630.8c

de Klerk GJ, Guan H, Huisman P, Marinova S (2011) Effects of phenolic compounds on adventitious root formation and oxidative decarboxylation of applied indoleacetic acid in *Malus* 'Jork 9'. Plant Growth Regul 63:175–185. https://doi.org/10.1007/s10725-010-9555-9

de Marcos LC, van Roermund CWT, Postis VLG et al (2013) Intrinsic acyl-CoA thioesterase activity of a peroxisomal ATP binding cassette transporter is required for transport and metabolism of fatty acids. Proc Natl Acad Sci 110:1279–1284. https://doi.org/10.1073/pnas.1218034110

de Oliveira DM, Finger-Teixeira A, Rodrigues Mota T et al (2015) Ferulic acid: a key component in grass lignocellulose recalcitrance to hydrolysis. Plant Biotechnol J 13:1224–1232. https://doi.org/10.1111/pbi.12292

de Oliveira DM, Finger Teixeira A, de Freitas DL et al (2017) Phenolic compounds in plants: Implications for bioenergy. In: Buckeridge S, de Souza AP (eds) Advances of basic science for second generation bioethanol from sugarcane. Springer, Berlin, pp 39–52

de Paiva LB, Goldbeck R, dos Santos WD, Squina FM (2013) Ferulic acid and derivatives: molecules with potential application in the pharmaceutical field. Braz J Pharm Sci 49:395–411. https://doi.org/10.1590/S1984-82502013000300002

DeClue MS, Baldridge KK, Künzler DE et al (2005) Isochorismate pyruvate lyase: a pericyclic reaction mechanism? J Am Chem Soc 127:15002–15003. https://doi.org/10.1021/ja055871t

Dedonder A, Van Sumere CF (1971) The effect of phenolics and related compounds on the growth and the respiration of *Chlorella vulgaris*. Z Pflanzenphysiol 65:70–80

Delaney TP, Uknes S, Vernooij B et al (1994) A central role of salicylic acid in plant disease resistance. Science 266(5188):1247–1250. https://doi.org/10.1126/science.266.5188.1247

Demos EK, Woolwine M, Wilson RH, McMillan C (1975) The effects of ten phenolic compounds on hypocotyl growth and mitochondrial metabolism of mung bean. Am J Bot 62:97–102. https://doi.org/10.2307/2442083

Dempsey DA, Vlot AC, Wildermuth MC, Klessig DF (2011) Salicylic acid biosynthesis and metabolism. Arabidopsis Book 9:e0156. https://doi.org/10.1199/tab.0156

Devi RS, Prasad MNV (1996) Ferulic acid mediated changes in oxidative enzymes of maize seedlings: implications in growth. Biol Plant 38:387–395. https://doi.org/10.1007/BF02896668

Dieryckx C, Gaudin V, Dupuy JW et al (2015) Beyond plant defense: insights on the potential of salicylic and methylsalicylic acid to contain growth of the phytopathogen *Botrytis cinerea*. Front Plant Sci 6:959. https://doi.org/10.3389/fpls.2015.00859

Ding J, Sun Y, Xiao CL et al (2007a) Physiological basis of different allelopathic reactions of cucumber and figleaf gourd plants to cinnamic acid. J Exp Bot 58:3765–3773. https://doi.org/10.1093/jxb/erm227

Ding L, Hofius D, Hajirezaei MR et al (2007b) Functional analysis of the essential bifunctional tobacco enzyme 3-dehydroquinate dehydratase/shikimate dehydrogenase in transgenic tobacco plants. J Exp Bot 58:2053–2067. https://doi.org/10.1093/jxb/erm059

Djordjevic M, Rolfe B (1988) Recognition process in the Rhizobium trifolii-white clover symbiosis. In: Bothe H, Brujin F, Newton F (eds) Nitrogen fixation: hundred years after. Springer, Stuttgart, pp 431–436

Doornbos RF, Geraats BPJ, Kuramae EE et al (2011) Effects of jasmonic acid, ethylene, and salicylic acid signaling on the rhizosphere bacterial community of *Arabidopsis thaliana*. Mol Plant Microbe Interact 24:395–407. https://doi.org/10.1094/MPMI-05-10-0115

dos Santos W, Ferrarese MLL, Ferrarese-Filho O (2008a) Ferulic acid: an allelochemical troublemaker. Funct Plant Sci Biotechnol 2:47–55

dos Santos WD, Ferrarese MDLL, Finger A et al (2004) Lignification and related enzymes in *Glycine max* root growth-inhibition by ferulic acid. J Chem Ecol 30:1203–1212. https://doi.org/10.1023/B:JOEC.0000030272.83794.f0

dos Santos WD, Ferrarese MLL, Nakamura CV et al (2008b) Soybean (*Glycine max*) root lignification induced by ferulic acid. The possible mode of action. J Chem Ecol 34:1230–1241. https://doi.org/10.1007/s10886-008-9522-3

dos Santos WD, Marchiosi R, Vilar FCM et al (2014) Polyvalent lignin: recent approaches in determination and applications. In: Lu F (ed) Lignin: structural analysis, applications in biomaterials and ecological significance. Nova Science, New York, pp 1–25

Duke SO (1985) Effects of herbicides on nonphotosynthetic biosynthetic processes. In: Duke SO (ed) Weed physiology, 1st edn. CRC Press, Boca Raton, pp 91–113

Durrant WE, Dong X (2004) Systemic acquired resistance. Annu Rev Phytopathol 42:185–209. https://doi.org/10.1146/annurev.phyto.42.040803.140421

Effmert U, Saschenbrecker S, Ross J et al (2005) Floral benzenoid carboxyl methyltransferases: from in vitro to in planta function. Phytochemistry 66:1211–1230. https://doi.org/10.1016/j.phytochem.2005.03.031

Einhellig FA (2004) Mode of allelochemical action of phenolic compounds. In: Macías FA, Galindo JCG, Molinillo JMG, Cutler HG (eds) Allelopathy—chemistry and mode of action allelochemicals. CRC Press, New York, pp 217–233

El-Basyouni SZ, Chen D, Ibrahim RK et al (1964) The biosynthesis of hydroxybenzoic acids in higher plants. Phytochemistry 3:485–492. https://doi.org/10.1016/S0031-9422(00)88025-5

El-Tayeb MA (2005) Response of barley grains to the interactive effect of salinity and salicylic acid. Plant Growth Regul 45:215–224. https://doi.org/10.1007/s10725-005-4928-1

Esmaeili M, Heidarzade A, Pirdashti H (2012) Inhibitory activity of pure allelochemicals on Barnyardgrass (*Echinochloa crus-galli* L.) seed and seedling parameters. Int J Agric Crop Sci 4(6):274–279

Faraji M, Fonseca LL, Escamilla-Treviño L et al (2018) Mathematical models of lignin biosynthesis. Biotechnol Biofuels 11:1–17. https://doi.org/10.1186/s13068-018-1028-9

Fariduddin Q, Hayat S, Ahmad A (2003) Salicylic acid influences net photosynthetic rate, carboxylation efficiency, nitrate reductase activity, and seed yield in *Brassica juncea*. Photosynthetica 41:281–284. https://doi.org/10.1023/B:PHOT.0000011962.05991.6c

Ferrarese MLL, Rodrigues JD, Ferrarese-Filho O (2000) Phenylalanine ammonia-lyase activity in soybean roots extract measured by reverse-phase high performance liquid chromatography. Plant Biol 2:152–153. https://doi.org/10.1055/s-2000-9162

Forouhar F, Yang Y, Kumar D et al (2005) Structural and biochemical studies identify tobacco SABP2 as a methyl salicylate esterase and implicate it in plant innate immunity. Proc Natl Acad Sci 102:1773–1778. https://doi.org/10.1073/pnas.0409227102

Funk C, Brodelius P (1990a) Phenylpropanoid metabolism in suspension cultures of *Vanilla planifolia* Andr. II Effects of precursor feeding and metabolic inhibitors. Plant Physiol 94:95–101. https://doi.org/10.1016/0031-9422(90)80030-K

Funk C, Brodelius PE (1990b) Phenylpropanoid metabolism in suspension cultures of *Vanilla planifolia* andr.: III. Conversion of 4-methoxycinnamic acids into 4-hydroxybenzoic acids. Plant Physiol 94:102–108. https://doi.org/10.1104/pp.94.1.102

Gaffney T, Friedrich L, Vernooij B et al (1993) Requirement of salicylic acid for the induction of systemic acquired resistance. Science 261(5122):754–756. https://doi.org/10.1126/science.261.5122.754

Gallage NJ, Hansen EH, Kannangara R et al (2014) Vanillin formation from ferulic acid in *Vanilla planifolia* is catalysed by a single enzyme. Nat Commun 5:4037. https://doi.org/10.1038/ncomms5037

Gallage NJ, Jørgensen K, Janfelt C et al (2018) The intracellular localization of the vanillin biosynthetic machinery in pods of *Vanilla planifolia*. Plant Cell Physiol 59:304–318. https://doi.org/10.1093/pcp/pcx185

Gallage NJ, Møller BL (2015) Vanillin-bioconversion and bioengineering of the most popular plant flavor and its de novo biosynthesis in the vanilla orchid. Mol Plant 8:40–57. https://doi.org/10.1016/j.molp.2014.11.008

Gasson MJ, Kitamura Y, McLauchlan WR et al (1998) Metabolism of ferulic acid to vanillin. J Biol Chem 273:4163–4170. https://doi.org/10.1074/jbc.273.7.4163

Gebhardt K (1982) Activation of indole-3 acetic acid oxidase from horseradish and *Prunus* by phenols and H_2O_2. Plant Growth Regul 1:73–84. https://doi.org/10.1007/BF00024500

Ghareib HRA, Abdelhamed MS, Ibrahim OH (2010) Antioxidative effects of the acetone fraction and vanillic acid from *Chenopodium murale* on tomato plants. Weed Biol Manag 10:64–72. https://doi.org/10.1111/j.1445-6664.2010.00368.x

Ghosh S, Chisti Y, Banerjee UC (2012) Production of shikimic acid. Biotechnol Adv 30:1425–1431. https://doi.org/10.1016/j.biotechadv.2012.03.001

Gosch C, Halbwirth H, Stich K (2010) Phloridzin: biosynthesis, distribution and physiological relevance in plants. Phytochemistry 71:838–843. https://doi.org/10.1016/j.phytochem.2010.03.003

Grabber JH, Schatz PF, Kim H et al (2010) Identifying new lignin bioengineering targets: 1. Monolignol-substitute impacts on lignin formation and cell wall fermentability. BMC Plant Biol 10:114. https://doi.org/10.1186/1471-2229-10-114

Greeland DJ (1971) Interactions between humic and fulvic acids and clays. Soil Sci 111:34–41

Guo J, Carrington Y, Alber A, Ehlting J (2014) Molecular characterization of quinate and shikimate metabolism in *Populus trichocarpa*. J Biol Chem 289:23846–23858. https://doi.org/10.1074/jbc.M114.558536

Halestrap AP (2009) What is the mitochondrial permeability transition pore? J Mol Cell Cardiol 46:821–831. https://doi.org/10.1016/j.yjmcc.2009.02.021

Hamada K, Tsutsumi Y, Nishida T (2003) Treatment of poplar callus with ferulic and sinapic acids II: effects on related monolignol biosynthetic enzyme activities. J Wood Sci 49:366–370. https://doi.org/10.1007/s10086-002-0475-9

Hammerbacher A, Coutinho TA, Gershenzon J (2019) Roles of plant volatiles in defense against microbial pathogens and microbial exploitation of volatiles. Plant Cell Environ. https://doi.org/10.1111/pce.13602

Hatayama T, Takeno K (2003) The metabolic pathway of salicylic acid rather than of chlorogenic acid is involved in the stress-induced flowering of *Pharbitis nil*. J Plant Physiol 160:461–467. https://doi.org/10.1078/0176-1617-01041

Hättenschwiler S, Vitousek PM (2000) The role of polyphenols in terrestrial ecosystem nutrient cycling. Tree 15:238–243. https://doi.org/10.1016/S0169-5347(00)01861-9

Havkin-Frenkel D, Podstolski A (2007) Vanillin production. USA patent, US2007/0231864 A1

Havkin-Frenkel D, Podstolski D, Dixon RA (2003) Vanillin biosynthetic pathway enzyme from *Vanilla planifolia*. USA patent, US2003/0070188 A1

Havkin-Frenkel D, Zylstra G, Frenkel C, Belanger F (2006) Production of vanillin in microbial cells. US patent, US20060172402 A1

Hayat Q, Hayat S, Irfan M, Ahmad A (2010) Effect of exogenous salicylic acid under changing environment: a review. Environ Exp Bot 68:14–25. https://doi.org/10.1016/j.envexpbot.2009.08.005

Herrmann KM (1995) The shikimate pathway: early steps in the biosynthesis of aromatic compounds. Plant Cell 7:907–919. https://doi.org/10.2307/3870046

Herrmann KM, Weaver LM (1999) The shikimate pathway. Annu Rev Plant Physiol Plant Mol Biol 50:473–503. https://doi.org/10.1146/annurev.arplant.50.1.473

Holopainen JK, Blande JD (2012) Molecular plant volatile communication. Adv Exp Med Biol 739:17–31. https://doi.org/10.1007/978-1-4614-1704-0_2

Hua J, Meyerowitz EM (1998) Ethylene responses are negatively regulated by a receptor gene family in *Arabidopsis thaliana*. Cell 94:261–271. https://doi.org/10.1016/S0092-8674(00)81425-7

Huang X, Bie Z (2010) Cinnamic acid-inhibited ribulose-1,5-bisphosphate carboxylase activity is mediated through decreased spermine and changes in the ratio of polyamines in cowpea. J Plant Physiol 167:47–53. https://doi.org/10.1016/j.jplph.2009.07.002

Huang X, Zhu G, Liu Q et al (2018) Modulation of plant salicylic acid-associated immune responses via glycosylation of dihydroxybenzoic acids. Plant Physiol 176:3103–3119. https://doi.org/10.1104/pp.17.01530

Huang YF, Chen CT, Kao CH (1993) Salicylic acid inhibits the biosynthesis of ethylene in detached rice leaves. Plant Growth Regul 12:79–82. https://doi.org/10.1007/BF00144586

Hunt L, Holdsworth MJ, Gray JE (2007) Nicotinamidase activity is important for germination. Plant J 51:341–351. https://doi.org/10.1111/j.1365-313X.2007.03151.x

Hussain MI, González L, Reigosa MJ (2010) Phytotoxic effects of allelochemicals and herbicides on photosynthesis, growth and carbon isotope discrimination in *Lactuca sativa*. Allelopath J 26:157–174

Hussain MI, Reigosa MJ (2011) Allelochemical stress inhibits growth, leaf water relations, PSII photochemistry, non-photochemical fluorescence quenching, and heat energy dissipation in three C_3 perennial species. J Exp Bot 62(13):4533–4545. https://doi.org/10.1093/jxb/err161

Hussain MS, Fareed S, Ansari S et al (2012) Current approaches toward production of secondary plant metabolites. J Pharm Bioallied Sci 4:10–20. https://doi.org/10.4103/0975-7406.92725

Igiehon NO, Babalola OO (2018) Below-ground-above-ground plant-microbial interactions: focusing on soybean, rhizobacteria and mycorrhizal fungi. Open Microbiol J 12:261–279. https://doi.org/10.2174/1874285801812010261

Inderjit M, Bhowmik PC (2004) Sorption of benzoic acid onto soil colloids and its implications for allelopathy studies. Biol Fertil Soils 40:345–348. https://doi.org/10.1007/s00374-004-0785-8

Ishihara T, Sekine KT, Hase S et al (2008) Overexpression of the *Arabidopsis thaliana* EDS5 gene enhances resistance to viruses. Plant Biol 10:451–461. https://doi.org/10.1111/j.1438-8677.2008.00050.x

Ishii-Iwamoto EL, Abrahim D, Sert MA et al (2006) Mitochondria as a site of allelochemical action. In: Reigosa MJ, Pedrol N, González L (eds) Allelopathy: a physiological process with ecological implications. Springer, Dordrecht, pp 267–284

James DG, Price TS (2004) Field-testing of methyl salicylate for recruitment and retention of beneficial insects in grapes and hops. J Chem Ecol 30:1613–1628

Jarvis AP, Schaaf O, Oldham NJ (2000) 3-Hydroxy-3-phenylpropanoic acid is an intermediate in the biosynthesis of benzoic acid and salicylic acid but benzaldehyde is not. Planta 212:119–126. https://doi.org/10.1007/s004250000377

Jazayeri O, Aghajanzadeh TA, Gildeh BS (2007) Study of growth factors, α-amylase and peroxidase activity in various cultivars of rice (*Oryza sativa* L.) under vanillic acid stress. Pakistan J Biol Sci 10:1673–1678

Ji Y, Liu J, Xing D (2016) Low concentrations of salicylic acid delay methyl jasmonate-induced leaf senescence by up-regulating nitric oxide synthase activity. J Exp Bot 67:5233–5245. https://doi.org/10.1093/jxb/erw280

Kennedy DO, Wightman EEL (2011) Herbal extracts and phytochemicals: plant secondary metabolites and the enhancement of human brain function. Adv Nutr 2:32–50. https://doi.org/10.3945/an.110.000117.32

Khalil S, Pawelek PD (2011) Enzymatic adenylation of 2,3-dihydroxybenzoate is enhanced by a protein-protein interaction between *Escherichia coli* 2,3-dihydro-2,3-dihydroxybenzoate dehydrogenase (EntA) and 2,3-dihydroxybenzoate-AMP ligase (EntE). Biochemistry 50:533–545. https://doi.org/10.1021/bi101558v

Khan W, Prithiviraj B, Smith DL (2003) Photosynthetic responses of corn and soybean to foliar application of salicylates. J Plant Physiol 160:485–492. https://doi.org/10.1078/0176-1617-00865

Kitzing K, Auweter S, Amrhein N, Macheroux P (2004) Mechanism of chorismate synthase: role of the two invariant histidine residues in the active site. J Biol Chem 279:9451–9461. https://doi.org/10.1074/jbc.M312471200

Kluge RA, Picoli AA, del Aguila JS (2010) Respiratory rate and ethylene production of whole and minimally processed beet roots submitted to ethylene and bioregulators treatments. Hortic Bras 28:54–57

Kobayashi K (2015) Plant methyl salicylate induces defense responses in the rhizobacterium *Bacillus subtilis*. Environ Microbiol 17:1365–1376. https://doi.org/10.1111/1462-2920.12613

Kundu A (2017) Vanillin biosynthetic pathways in plants. Planta 245:1069–1078. https://doi.org/10.1007/s00425-017-2684-x

Kutchan TM, Gershenzon J, Moller BL, Gang DR (2015) Natural products. In: Buchanan BB, Gruissem W, Jones RL (eds) Biochemistry & molecular biology of plants. Wiley, New York, pp 1132–1205

Lam PY, Liu H, Lo C (2015) Completion of tricin biosynthesis pathway in rice: cytochrome P450 75B4 is a unique chrysoeriol 5′-hydroxylase. Plant Physiol 168:1527–1536. https://doi.org/10.1104/pp.15.00566

Lan W, Lu F, Regner M et al (2015) Tricin, a flavonoid monomer in monocot lignification. Plant Physiol 167:1284–1295. https://doi.org/10.1104/pp.114.253757

Lebeis SL, Paredes SH, Lundberg DS et al (2015) Salicylic acid modulates colonization of the root microbiome by specific bacterial taxa. Science 349(6250):860–864. https://doi.org/10.5061/dryad.238b2

Lee RB (1977) Effects of organic acids on the loss of ions from barley roots. J Exp Bot 28:578–587. https://doi.org/10.1093/jxb/28.3.578

León J, Shulaev V, Yalpani N et al (1995) Benzoic acid 2-hydroxylase, a soluble oxygenase from tobacco, catalyzes salicylic acid biosynthesis. Proc Natl Acad Sci

92:10413–10417. https://doi.org/10.1073/pnas.92.22.10413

Leslie CA, Romani RJ (1988) Inhibition of ethylene biosynthesis by salicylic acid. Plant Physiol 88:833–837. https://doi.org/10.1104/pp.88.3.833

Li HH, Inoue M, Nishimura H et al (1993) Interactions of *trans*-cinnamic acid, its related phenolic allelochemicals, and abscisic acid in seedling growth and seed germination of lettuce. J Chem Ecol 19:1775–1787. https://doi.org/10.1007/BF00982307

Li ZH, Wang Q, Ruan X et al (2010) Phenolics and plant allelopathy. Molecules 15:8933–8952. https://doi.org/10.3390/molecules15128933

Lima RB, Salvador VH, dos Santos WD et al (2013) Enhanced lignin monomer production caused by cinnamic acid and its hydroxylated derivatives inhibits soybean root growth. PLoS ONE 8:1–8. https://doi.org/10.1371/journal.pone.0080542

Lopez-Vergara HI, Jackson KM, Meshesha LZ, Metrik J (2019) Dysregulation as a correlate of cannabis use and problem use. Addict Behav 95:138–144. https://doi.org/10.1016/j.addbeh.2019.03.010

Lu XF, Zhang H, Lyu SS et al (2018) Effects of exogenous phenolic acids on photosystem functions and photosynthetic electron transport rate in strawberry leaves. Photosynthetica 56:616–622. https://doi.org/10.1007/s11099-017-0702-7

Lugtenberg B, Kamilova F (2009) Plant-growth-promoting rhizobacteria. Annu Rev Microbiol 63:541–556. https://doi.org/10.1016/B978-0-12-374984-0.01169-4

Macheroux P, Schmid J, Amrhein N, Schaller A (1999) A unique reaction in a common pathway: mechanism and function of chorismate synthase in the shikimate pathway. Planta 207:325–334. https://doi.org/10.1007/s004250050489

Macías FA, Mejías FJR, Molinillo JMG (2019) Recent advances in allelopathy for weed control: from knowledge to applications. Pest Manag Sci 75:2413–2436

Maeda H, Dudareva N (2012) The shikimate pathway and aromatic amino acid biosynthesis in plants. Annu Rev Plant Biol 63:73–105. https://doi.org/10.1146/annurev-arplant-042811-105439

Makovec P, Sindelár L (1984) The effect of phenolic compounds on the activity of respiratory chain enzymes and on respiration and phosphorylation activities of potato tuber mitochondria. Biol Plant 26:415–422. https://doi.org/10.1007/BF02909590

Mallinger RE, Hogg DB, Gratton C (2011) Methyl salicylate attracts natural enemies and reduces populations of soybean aphids (Hemiptera: Aphididae) in soybean agroecosystems. J Econ Entomol 104:115–124. https://doi.org/10.1603/EC10253

Mandal S, Mandal M, Das A et al (2009) Stimulation of indoleacetic acid production in a *Rhizobium* isolate of *Vigna mungo* by root nodule phenolic acids. Arch Microbiol 191:389–393. https://doi.org/10.1007/s00203-008-0455-6

Mandal SM, Chakraborty D, Dey S (2010) Phenolic acids act as signaling molecules in plant-microbe symbioses. Plant Signal Behav 5:359–368. https://doi.org/10.4161/psb.5.4.10871

Martí S, Andrés J, Moliner V et al (2009) Mechanism and plasticity of isochorismate pyruvate lyase: a computational study. J Am Chem Soc 131:16156–16161. https://doi.org/10.1021/ja905271g

Martínez C, Pons E, Prats G, León J (2004) Salicylic acid regulates flowering time and links defense responses and reproductive development. Plant J 37:209–217. https://doi.org/10.1046/j.1365-313X.2003.01954.x

Martini X, Pelz-Stelinski KS, Stelinski LL (2014) Plant pathogen-induced volatiles attract parasitoids to increase parasitism of an insect vector. Front Ecol Evol 2:1–8. https://doi.org/10.3389/fevo.2014.00008

Mittelstädt G, Negron L, Schofield LR et al (2013) Biochemical and structural characterisation of dehydroquinate synthase from the New Zealand kiwifruit *Actinidia chinensis*. Arch Biochem Biophys 537:185–191. https://doi.org/10.1016/j.abb.2013.07.022

Moreland DE, Novitzky WP (1987) Effects of phenolic acids, coumarins, and flavonoids on isolated chloroplasts and mitochondria. In: Waller GR (ed) Allelochemicals: role in agriculture and forestry. American Chemical Society, Washington, DC, pp 247–261

Mittova V, Tal M, Volokita M, Guy M (2003) Up-regulation of the leaf mitochondrial and peroxisomal antioxidative systems in response to salt-induced oxidative stress in the wild salt-tolerant tomato species *Lycopersicon pennellii*. Plant Cell Environ 26:845–856. https://doi.org/10.1046/j.1365-3040.2003.01016.x

Moreno PRH, Van Der HR, Verpoorte R (1996) Elicitor-mediated induction of isochorismate synthase and accumulation of 2,3-dihydroxybenzoic acid *Catharanthus roseus* cell suspension and shoot cultures. Plant Cell Rep 15:516–521

Muir RM, Ibáñez AM, Uratsu SL et al (2011) Mechanism of gallic acid biosynthesis in bacteria (*Escherichia coli*) and walnut (*Juglans regia*). Plant Mol Biol 75:555–565. https://doi.org/10.1007/s11103-011-9739-3

Mustafa NR, Kim HK, Choi YH et al (2009) Biosynthesis of salicylic acid in fungus elicited *Catharanthus roseus* cells. Phytochemistry 70:532–539. https://doi.org/10.1016/j.phytochem.2009.01.009

Mustafa NR, Verpoorte R (2005) Chorismate derived C6C1 compounds in plants. Planta 222:1–5. https://doi.org/10.1007/s00425-005-1554-0

Naczk M, Shahidi F (2006) Phenolics in cereals, fruits and vegetables: occurrence, extraction and analysis. J Pharm Biomed Anal 41:1523–1542. https://doi.org/10.1016/j.jpba.2006.04.002

Nair RB, Bastress KL, Ruegger MO et al (2004) The *Arabidopsis thaliana* reduced epidermal fluorescence1 gene encodes an aldehyde dehydrogenase involved in ferulic acid and sinapic acid biosynthesis. Plant Cell 16:544–554. https://doi.org/10.1105/tpc.017509

Nawrath C (2002) EDS5, an essential component of salicylic acid-dependent signaling for disease resistance in *Arabidopsis*, is a member of the MATE transporter family. Plant Cell 14:275–286. https://doi.org/10.1105/tpc.010376

Niggeweg R, Michael AJ, Martin C (2004) Engineering plants with increased levels of the antioxidant chlorogenic acid. Nat Biotechnol 22:746–754. https://doi.org/10.1038/nbt966

Norman C, Howell KA, Millar AH et al (2004) Salicylic acid is an uncoupler and inhibitor of mitochondrial electron transport. Plant Physiol 134:492–501. https://doi.org/10.1104/pp.103.031039

Ogo Y, Ozawa K, Ishimaru T et al (2013) Transgenic rice seed synthesizing diverse flavonoids at high levels: a new platform for flavonoid production with associated health benefits. Plant Biotechnol J 11:734–746. https://doi.org/10.1111/pbi.12064

Oliveira DM, Mota TR, Oliva B et al (2019) Feruloyl esterases: biocatalysts to overcome biomass recalcitrance and for the production of bioactive compounds. Bioresour Technol 278:408–423. https://doi.org/10.1016/j.biortech.2019.01.064

Pak FE, Gropper S, Dai WD et al (2004) Characterization of a multifunctional methyltransferase from the orchid *Vanilla planifolia*. Plant Cell Rep 22:959–966. https://doi.org/10.1007/s00299-004-0795-x

Park SW, Kaimoyo E, Kumar D et al (2007) Methyl salicylate is a critical mobile signal for plant systemic acquired resistance. Science 318:113–116. https://doi.org/10.1126/science.1147113

Parsons HL, Yip JYH, Vanlerberghe GC (1999) Increased respiratory restriction during phosphate-limited growth in transgenic tobacco cells lacking alternative oxidase. Plant Physiol 121:1309–1320. https://doi.org/10.1104/pp.121.4.1309

Peñuelas J, Ribas-Carbo M, Giles L (1996) Effects of allelochemicals on plant respiration and oxygen isotope fractionation by the alternative oxidase. J Chem Ecol 22:801–805

Petrik DL, Karlen SD, Cass CL et al (2014) *p*-Coumaroyl-CoA: monolignol transferase (PMT) acts specifically in the lignin biosynthetic pathway in *Brachypodium distachyon*. Plant J 77:713–726. https://doi.org/10.1111/tpj.12420

Pierpoint WS (1997) The natural history of salicylic acid plant product and mammalian medicine. Interdiscip Sci Rev 22:45–52. https://doi.org/10.1179/030801897789765129

Pieterse CMJ, Zamioudis C, Berendsen RL et al (2014) Induced systemic resistance by beneficial microbes. Annu Rev Phytopathol 52:347–375. https://doi.org/10.1146/annurev-phyto-082712-102340

Podstolski A, Havkin-Frenkel D, Malinowski J et al (2002) Unusual 4-hydroxybenzaldehyde synthase activity from tissue cultures of the vanilla orchid *Vanilla planifolia*. Phytochemistry 61:611–620. https://doi.org/10.1016/S0031-9422(02)00285-6

Polidoros AN, Mylona PV, Pasentsis K et al (2005) The maize alternative oxidase 1a (*Aox1a*) gene is regulated by signals related to oxidative stress. Redox Rep 10:71–78. https://doi.org/10.1179/135100005x21688

Popov VN, Simonian RA, Skulachev VP, Starkov AA (1997) Inhibition of the alternative oxidase stimulates H_2O_2 production in plant mitochondria. FEBS Lett 415:87–90. https://doi.org/10.1016/S0014-5793(97)01099-5

Poulev A, Heckman JR, Raskin I, Belanger FC (2019) Tricin levels and expression of flavonoid biosynthetic genes in developing grains of purple and brown pericarp rice. PeerJ 7:e6477. https://doi.org/10.7717/peerj.6477

Premoli M, Aria F, Bonini SA et al (2019) Cannabidiol: recent advances and new insights for neuropsychiatric disorders treatment. Life Sci 224:120–127. https://doi.org/10.1016/J.LFS.2019.03.053

Purohit S, Laloraya MM, Bharti S (1991) Effect of phenolic compounds on abscisic acid-induced stomatal movement: structure—activity relationship. Physiol Plant 81:79–82. https://doi.org/10.1111/j.1399-3054.1991.tb01716.x

Qualley AV, Widhalm JR, Adebesin F et al (2012) Completion of the core-oxidative pathway of benzoic acid biosynthesis in plants. Proc Natl Acad Sci 109:16383–16388. https://doi.org/10.1073/pnas.1211001109

Rajput P, Rao PB (2013) Weed management through bio-herbicidal activity of rice straw for mitigating herbicide dependency. Am J Agric Environ Sci 13:1533–1540. https://doi.org/10.5829/idosi.aejaes.2013.13.11.1812

Raskin I, Ehmann A, Melander WR, Meeuse BJD (1987) Salicylic acid: a natural inducer of heat production in *Arum* lilies. Science 237:1601–1602. https://doi.org/10.1126/science.237.4822.1601

Raskin I, Turner IM, Melander WR (1989) Regulation of heat production in the inflorescences of an *Arum* lily by endogenous salicylic acid. Proc Natl Acad Sci 86:2214–2218. https://doi.org/10.1073/pnas.86.7.2214

Rasmann S, Turlings TCJ (2016) Root signals that mediate mutualistic interactions in the rhizosphere. Curr Opin Plant Biol 32:62–68. https://doi.org/10.1016/j.pbi.2016.06.017

Saha B, Borovskii G, Panda SK (2016) Alternative oxidase and plant stress tolerance. Plant Signal Behav 11:e1256530. https://doi.org/10.1080/15592324.2016.1256530

Salvador VH, Lima RB, dos Santos WD et al (2013) Cinnamic acid increases lignin production and inhibits soybean root growth. PLoS ONE 8:1–10. https://doi.org/10.1371/journal.pone.0069105

Schmidt CL, Danneel H-J, Schultz G, Buchanan BB (1990) Shikimate kinase from spinach chloroplasts. Plant Physiol 93:758–766

Schönbrunn E, Eschenburg S, Shuttleworth WA et al (2001) Interaction of the herbicide glyphosate with its target enzyme 5-enolpyruvylshikimate 3-phosphate synthase in atomic detail. Proc Natl Acad Sci 98:1376–1380. https://doi.org/10.1073/pnas.98.4.1376

Seneviratne G, Jayasinghearachchi HS (2003) Phenolic acids: possible agents of modifying N_2-fixing symbiosis through rhizobial alteration? Plant Soil 252:385–395. https://doi.org/10.1023/A:1024725511783

Sert MA, Ferrarese MLL, Bernadelli YR et al (1997) Effects of ferulic acid on L-malate oxidation in isolated soybean mitochondria. Biol Plant 40:345–350

Sharma P, Jha AB, Dubey RS, Pessarakli M (2012) Reactive oxygen species, oxidative damage, and antioxidative defense mechanism in plants under stressful conditions. J Bot 2012:1–26. https://doi.org/10.1155/2012/217037

Sharma A, Sidhu GPS, Araniti F, Bali AS, Shahzad B, Tripathi DK, Brestic M, Skalicky M, Landi M (2020) The role of salicylic acid in plants exposed to heavy metals. Molecules 25:540–561. https://doi.org/10.3390/molecules25030540

Shin SY, Kim IS, Kim YS et al (2013) Ectopic expression of *Brassica rapa* L. MDHAR increased tolerance to freezing stress by enhancing antioxidant systems of host plants. S Afr J Bot 88:388–400. https://doi.org/10.1016/j.sajb.2013.08.015

Shitan N (2016) Secondary metabolites in plants: transport and self-tolerance mechanisms. Biosci Biotechnol Biochem 80:1283–1293. https://doi.org/10.1080/09168451.2016.1151344

Singh HP, Kaur S, Batish DR, Kohli RK (2009) Caffeic acid inhibits in vitro rooting in mung bean [*Vigna radiata* (L.) Wilczek] hypocotyls by inducing oxidative stress. Plant Growth Regul 57:21–30. https://doi.org/10.1007/s10725-008-9314-3

Singh PK, Singh R, Singh S (2013) Cinnamic acid induced changes in reactive oxygen species scavenging enzymes and protein profile in maize (*Zea mays* L.) plants grown under salt stress. Physiol Mol Biol Plants 19:53–59. https://doi.org/10.1007/s12298-012-0126-6

Singh SA, Christendat D (2006) Structure of *Arabidopsis* dehydroquinate dehydratase-shikimate dehydrogenase and implications for metabolic channeling in the shikimate pathway. Biochemistry 45:7787–7796. https://doi.org/10.1021/bi060366

Singh SA, Christendat D (2007) The DHQ-dehydroshikimate-SDH-shikimate-NADP(H) complex: insights into metabolite transfer in the shikimate pathway. Cryst Growth Des 7:2153–2160. https://doi.org/10.1021/cg7007107

Smith LM (2019) Salicylic acid, senescence, and heterosis. Plant Physiol 180:3–4. https://doi.org/10.1104/pp.19.00260

Snoeren TAL, Mumm R, Poelman EH et al (2010) The herbivore-induced plant volatile methyl salicylate negatively affects attraction of the parasitoid *Diadegma semiclausum*. J Chem Ecol 36:479–489. https://doi.org/10.1007/s10886-010-9787-1

Stallings WC, Abdel-Meguid SS, Lim LW et al (1991) Structure and topological symmetry of the glyphosate target 5-enolpyruvylshikimate-3-phosphate synthase: a distinctive protein fold. Proc Natl Acad Sci 88:5046–5050. https://doi.org/10.1073/pnas.88.11.5046

Staswick PE, Su W, Howell SH (1992) Methyl jasmonate inhibition of root growth and induction of a leaf protein are decreased in an *Arabidopsis thaliana* mutant. Proc Natl Acad Sci 89:6837–6840. https://doi.org/10.1073/pnas.89.15.6837

Stepanycheva EA, Petrova MO, Chermenskaya TD, Shamshev IV (2016) Effect of methyl salicylate on behavioral responses of insects in a forest park. Entomol Rev 96:284–287. https://doi.org/10.1134/S0013873816030052

Stevens J, Senaratna T, Sivasithamparam K (2006) Salicylic acid induces salinity tolerance in tomato (*Lycopersicon esculentum* cv. Roma): associated changes in gas exchange, water relations and membrane stabilisation. Plant Growth Regul 49:77–83. https://doi.org/10.1007/s10725-006-0019-1

Sun WJ, Nie YX, Gao Y et al (2012) Exogenous cinnamic acid regulates antioxidant enzyme activity and reduces lipid peroxidation in drought-stressed cucumber leaves. Acta Physiol Plant 34:641–655. https://doi.org/10.1007/s11738-011-0865-y

Takeoka TN (1968) Formation of protocatechuic acid from 5-dehydroshikimic acid in the extract of mung bean seedling. Bot Mag Tokyo 81:103–104

Tharayil N, Bhowmik PC, Xing B (2006) Preferential sorption of phenolic phytotoxins to soil: implications for altering the availability of allelochemicals. J Agric Food Chem 54:3033–3040. https://doi.org/10.1021/jf053167q

Tobimatsu Y, Elumalai S, Grabber JH et al (2012) Hydroxycinnamate conjugates as potential monolignol replacements: in vitro lignification and cell wall studies with rosmarinic acid. Chemsuschem 5:676–686. https://doi.org/10.1002/cssc.201100573

Tohge T, Watanabe M, Hoefgen R, Fernie AR (2013) Shikimate and phenylalanine biosynthesis in the green lineage. Front Plant Sci 4:1–13. https://doi.org/10.3389/fpls.2013.00062

Trezzi MM, Vidal RA, Junior AAB et al (2016) Allelopathy: driving mechanisms governing its activity in agriculture. J Plant Interact 11:53–60. https://doi.org/10.1080/17429145.2016.1159342

Tsao R, Deng Z (2004) Separation procedures for naturally occurring antioxidant phytochemicals. J Chromatogr B 812:85–99. https://doi.org/10.1016/j.jchromb.2004.09.028

Tzin V, Galili G (2010a) New insights into the shikimate and aromatic amino acids biosynthesis pathways in plants. Mol Plant 3:956–972. https://doi.org/10.1093/mp/ssq048

Tzin V, Galili G (2010) The biosynthetic pathways for shikimate and aromatic amino acids in Arabidopsis thaliana. In: Torii K (ed) The Arabidopsis book. American Society of Plant Biologists, Rockville, p 8

Umezawa T (2018) Lignin modification in planta for valorization. Phytochem Rev 17:1305–1327. https://doi.org/10.1007/s11101-017-9545-x

van Overbeek J, Blondeau R, Horne V (1951) Trans-cinnamic acid as an anti-auxin. Am J Bot 38:589–595. https://doi.org/10.2307/2437768

Vanholme R, Cesarino I, Rataj K et al (2013) Caffeoyl shikimate esterase (CSE) is an enzyme in the lignin biosynthetic pathway in *Arabidopsis*. Science 341:1103–1106. https://doi.org/10.1126/science.1241602

Vanholme R, Demedts B, Morrel K et al (2010) Lignin biosynthesis and structure. Plant Physiol 153:895–905. https://doi.org/10.1104/pp.110.155119

Veach AM, Morris R, Yip DZ et al (2019) Rhizosphere microbiomes diverge among *Populus trichocarpa* plant-host genotypes and chemotypes, but it depends on soil origin. Microbiome 7:1–15. https://doi.org/10.1186/s40168-019-0668-8

Venturi V, Keel C (2016) Signaling in the rhizosphere. Trends Plant Sci 21:187–198. https://doi.org/10.1016/j.tplants.2016.01.005

Verberne MC, Verpoorte R, Bol JF et al (2000) Overproduction of salicylic acid in plants by bacterial transgenes enhances pathogen resistance. Nat Biotechnol 18:779–783. https://doi.org/10.1038/77347

Vernooij B, Friedrichya L, Reist R et al (1994) Salicylic acid is not the translocated signal responsible for inducing systemic acquired resistance but is required in signal transduction. Plant Cell 6:959–965. https://doi.org/10.1105/tpc.6.7.959

Wan YY, Chen SY, Huang YW et al (2014) Caffeic acid pretreatment enhances dehydration tolerance in cucumber seedlings by increasing antioxidant enzyme activity and proline and soluble sugar contents. Sci Hortic 173:54–64. https://doi.org/10.1016/j.scienta.2014.04.033

Wang SP, Lin SC, Li S et al (2016) Potent antiarthritic properties of phloretin in murine collagen-induced arthritis.

Evid Based Complem Altern Med 2016:1–9. https://doi.org/10.1155/2016/9831263
Wang XQ, Du GD, Lu XF et al (2019) Characteristics of mitochondrial membrane functions and antioxidant enzyme activities in strawberry roots under exogenous phenolic acid stress. Sci Hortic 248:89–97. https://doi.org/10.1016/j.scienta.2018.12.051
War AR, Paulraj MG, Ahmad T et al (2012) Mechanisms of plant defense against insect herbivores. Plant Signal Behav 7:1306–1320. https://doi.org/10.4161/psb.21663
White RF (1979) Acetylsalicylic acid (aspirin) induces resistance to tobacco mosaic virus in tobacco. Virology 99:410–412. https://doi.org/10.1016/0042-6822(79)90019-9
Whitehead DC (1964) Identification of *p*-hydroxybenzoic, vanillic, *p*-coumaric and ferulic acids in soils. Nature 202:417–418
Widhalm JR, Dudareva N (2015) A familiar ring to it: biosynthesis of plant benzoic acids. Mol Plant 8:83–97. https://doi.org/10.1016/j.molp.2014.12.001
Wildermuth MC (2006) Variations on a theme: synthesis and modification of plant benzoic acids. Curr Opin Plant Biol 9:288–296. https://doi.org/10.1016/j.pbi.2006.03.006
Wildermuth MC, Dewdney J, Wu G, Ausubel FM (2001) Isochorismate synthase is required to synthesize salicylic acid for plant defense. Nature 414:562–565. https://doi.org/10.1038/35107108
Wilson PJ, Van Staden SJ (1990) Rhizocaline, rooting co-factors, and the concept of promoters and inhibitors of adventitious rooting—a review. Ann Bot 66:479–490. https://doi.org/10.1093/oxfordjournals.aob.a088051
Wink M (2018) Plant secondary metabolites modulate insect behavior-steps toward addiction? Front Physiol 9:1–9. https://doi.org/10.3389/fphys.2018.00364
Wink M (2015) Modes of action of herbal medicines and plant secondary metabolites. Medicines 2:251–286. https://doi.org/10.3390/medicines2030251
Wink M (2010) Biochemistry, physiology and ecological functions of secondary metabolites. In: Wink M (ed) Biochemistry of plant secondary metabolism, 2nd edn. Blackwell, Hoboken
Withers S, Lu F, Kim H et al (2012) Identification of grass-specific enzyme that acylates monolignols with *p*-coumarate. J Biol Chem 287:8347–8355. https://doi.org/10.1074/jbc.m111.284497
Yamauchi K, Fukushima K (2004) The regulation from guaiacyl to syringyl lignin in the differentiating xylem of *Robinia pseudoacacia*. C R Biol 327:791–797. https://doi.org/10.1016/j.crvi.2004.07.012
Yang CM, Chang IF, Lin SJ, Chou CH (2004) Effects of three allelopathic phenolics on chlorophyll accumulation of rice (*Oryza sativa*) seedlings: II. Stimulation of consumption-orientation. Bot Bull Acad Sin 45:119–125
Yang CM, Lee CN, Chou CH (2002) Effects of three allelopathic phenolics on chlorophyll accumulation of rice (*Oryza sativa*) seedlings: I. Inhibition of supply-orientation. Bot Bull Acad Sin 43:299–304
Yang H, Barros-rios J, Kourteva G et al (2017) A re-evaluation of the final step of vanillin biosynthesis in the orchid *Vanilla planifolia*. Phytochemistry 139:33–46. https://doi.org/10.1016/j.phytochem.2017.04.003
Ye SF, Yu JQ, Peng YH et al (2004) Incidence of Fusarium wilt in *Cucumis sativus* L. is promoted by cinnamic acid, an autotoxin in root exudates. Plant Soil 263:143–150. https://doi.org/10.1023/B:PLSO.0000047721.78555.dc
Yu JQ, Ye SF, Zhang MF, Hu WH (2003) Effects of root exudates and aqueous root extracts of cucumber (*Cucumis sativus*) and allelochemicals, on photosynthesis and antioxidant enzymes in cucumber. Biochem Syst Ecol 31:129–139. https://doi.org/10.1016/S0305-1978(02)00150-3
Zabalza A, Orcaray L, Fernández-Escalada M et al (2017) The pattern of shikimate pathway and phenylpropanoids after inhibition by glyphosate or quinate feeding in pea roots. Pestic Biochem Physiol 141:96–102. https://doi.org/10.1016/j.pestbp.2016.12.005
Zenk MH (1965) Biosynthese von vanillin in *Vanilla planifolia*. Andr Z Pflanzenphysiol 53:404
Zhang K, Halitschke R, Yin C et al (2013) Salicylic acid 3-hydroxylase regulates *Arabidopsis* leaf longevity by mediating salicylic acid catabolism. Proc Natl Acad Sci 110:14807–14812. https://doi.org/10.1073/pnas.1302702110
Zhang Y, Zhao L, Zhao J et al (2017) S5H/DMR6 encodes a salicylic acid 5-hydroxylase that fine-tunes salicylic acid homeostasis. Plant Physiol 175:1082–1093. https://doi.org/10.1104/pp.17.00695
Zheng X, Van Huystee RB (1992) Anionic peroxidase catalysed ascorbic acid and IAA oxidation in the presence of hydrogen peroxide: a defense system against peroxidative stress in peanut plant. Phytochemistry 31:1895–1898. https://doi.org/10.1016/0031-9422(92)80329-D
Zhou XM, MacKenzie AF, Madramootoo CA, Smith DL (1999) Effects of stem-injected plant growth regulators, with or without sucrose, on grain production, biomass and photosynthetic activity of field-grown corn plants. J Agron Crop Sci 183:103–110. https://doi.org/10.1046/j.1439-037X.1999.00331.x

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