



## Review



## Structural insights, biocatalytic characteristics, and application prospects of lignin-modifying enzymes for sustainable biotechnology

Anil Kumar Singh<sup>a,b</sup>, Hafiz M.N. Iqbal<sup>c</sup>, Nunzio Cardullo<sup>d</sup>, Vera Muccilli<sup>d</sup>,  
 Jesús Fernández-Lucas<sup>e,f</sup>, Jens Ejbye Schmidt<sup>g</sup>, Teofil Jesionowski<sup>h</sup>, Muhammad Bilal<sup>h,i,\*</sup>

<sup>a</sup> Environmental Microbiology Laboratory, Environmental Toxicology Group CSIR-Indian Institute of Toxicology Research (CSIR-IITR), Vishvighyan Bhawan, 31, Mahatma Gandhi Marg, Lucknow 226001, Uttar Pradesh, India

<sup>b</sup> Academy of Scientific and Innovative Research (AcSIR), Ghaziabad 201002, India

<sup>c</sup> Tecnológico de Monterrey, School of Engineering and Sciences, Monterrey 64849, Mexico

<sup>d</sup> Dipartimento di Scienze Chimiche, Università degli Studi di Catania, V.le A. Doria 6, 95125 Catania, Italy

<sup>e</sup> Applied Biotechnology Group, Universidad Europea de Madrid, Urbanización El Bosque, 28670, Villaviciosa de Odón, Spain

<sup>f</sup> Grupo de Investigación en Ciencias Naturales y Exactas, GICNEX, Universidad de la Costa, CUC, Calle 58 # 55-66, 080002, Barranquilla, Colombia

<sup>g</sup> Department of Green Technology, University of Southern Denmark, Campusvej 55, DK-5230 Odense M, Denmark

<sup>h</sup> Institute of Chemical Technology and Engineering, Faculty of Chemical Technology, Poznan University of Technology, Berdychowo 4, PL-60965 Poznan, Poland

<sup>i</sup> Department of Sanitary Engineering, Faculty of Civil and Environmental Engineering, Gdansk University of Technology, G. Narutowicza 11/12 Str., 80-233 Gdansk, Poland

## ARTICLE INFO

## Keywords:

Lignin modifying enzymes  
 Catalytic mechanisms  
 Depolymerization  
 Environmental pollutants  
 Computational framework  
 Sustainable mitigation

## ABSTRACT

Lignin modifying enzymes (LMEs) have gained widespread recognition in depolymerization of lignin polymers by oxidative cleavage. LMEs are a robust class of biocatalysts that include lignin peroxidase (LiP), manganese peroxidase (MnP), versatile peroxidase (VP), laccase (LAC), and dye-decolorizing peroxidase (DyP). Members of the LMEs family act on phenolic, non-phenolic substrates and have been widely researched for valorization of lignin, oxidative cleavage of xenobiotics and phenolics. LMEs implementation in the biotechnological and industrial sectors has sparked significant attention, although its potential future applications remain underexplored. To understand the mechanism of LMEs in sustainable pollution mitigation, several studies have been undertaken to assess the feasibility of LMEs in correlating to diverse pollutants for binding and intermolecular interactions at the molecular level. However, further investigation is required to fully comprehend the underlying mechanism. In this review we presented the key structural and functional features of LMEs, including the computational aspects, as well as the advanced applications in biotechnology and industrial research. Furthermore, concluding remarks and a look ahead, the use of LMEs coupled with computational framework, built upon artificial intelligence (AI) and machine learning (ML), has been emphasized as a recent milestone in environmental research.

## 1. Introduction

Lignin modifying enzymes (LMEs) have long been considered crucial enzymes that act on lignin polymers in a variety of ways, including catalytic cleavage of C-C, C-O-C bond, depolymerization, and valorization [1–7]. Taking advantage of their role in lignin degradation, LMEs have been used in a variety of biotechnological processes and applications including; pulp bleaching for paper manufacturing, food, textile, dye, cosmetics industries, and further sustainable generation of

renewable chemicals for organic synthesis, bio-materials, fuels, and bioremediation of hazardous pollutants [8–12]. The use of LMEs has significantly increased over the last two decades with the awareness of the importance of employing “green approaches” in the industry which are based on sustainable, environmentally friendly, economically viable, and potentially high-yielding processes without producing pollutants during reaction process [13–15]. Fig. 1 portrays a concise summary of LMEs and their respective functionalities in biotechnological and industrial sector. LMEs comprises a set of five different enzymes, which

\* Corresponding author at: Institute of Chemical Technology and Engineering, Faculty of Chemical Technology, Poznan University of Technology, Berdychowo 4, PL-60965 Poznan, Poland.

E-mail address: [muhammad.bilal@put.poznan.pl](mailto:muhammad.bilal@put.poznan.pl) (M. Bilal).

<https://doi.org/10.1016/j.ijbiomac.2023.124968>

Received 4 March 2023; Received in revised form 22 April 2023; Accepted 17 May 2023

Available online 20 May 2023

0141-8130/© 2023 The Author(s). Published by Elsevier B.V. This is an open access article under the CC BY-NC-ND license (<http://creativecommons.org/licenses/by-nc-nd/4.0/>).

similarly perform as an oxidoreductase in catalytic action: lignin peroxidase (LiP; EC 1.11.1.14), manganese peroxidase (MnP; EC 1.11.1.13), laccase (LAC; EC 1.10.3.2), versatile peroxidase (VP; EC 1.11.1.16), and dye decolorizing peroxidase (DyP; EC 1.11.1.19) [16–18]. LMEs member LAC contains multi copper in its protein structure hence called multi-copper oxidase (MCO) [19–21]. It acts on the phenolic substrate in presence of molecular oxygen ( $O_2$ ) [19]. However, LiP and DyP are heme proteins with Fe (III), while MnP possess Mn (II) centers, respectively [22–24]. Reaction mechanism of these oxidoreductase members except LAC are  $H_2O_2$ -dependent that act on a wide variety of phenolic and non-phenolic substrates in a similar fashion of LiP's catalytic cycle involving  $H_2O_2$  dependent, initiates a two-electron oxidation of a native LiP to first produce an oxoferryl iron radical cation intermediate, Compound I, along with the reduction of  $H_2O_2$  to water, and complete reaction in subsequent cycles [25,26]. Those lignin-degrading peroxidases are likely to be a member of the plant peroxidase superfamily (Class II) [27,28]. Among aforesaid, DyP has been recently identified class of heme-containing peroxidases and given the name DyP because it has an effective, lignin degradation, and dye decolorizing capability on a variety of industrial dyes [3,29–31]. Fungal species are well documented for secretion or production of LMEs in nature, including; *Phanerochaete chrysosporium*, *Phlebia floridensis*, *Trametes versicolor*, *Phlebia fascicularia*, *Dichomitus squalens*, *Daedalea flavida*, *Phlebia radiata*, *Cerrena unicolor*, *Trametes trogii* and many others [32–34]. Fungal LMEs work at weak acid to neutral pH (4–7) and diverse temperature range (30–55 °C) for the activity of the degradation and decolorization processes [35–37]. However, no single fungal species are known to produce all members of LMEs. The white-rot fungi (WRF) has largely been focused on acting as potential lignin degraders, including lignin, xenobiotic, and recalcitrant compounds via the extracellular  $H_2O_2$ -dependent oxidoreductase enzyme mechanism [34,38–42]. Several White-Rot Basidiomycete (WRB) have been examined in recent years to identify viable LMEs producers, while a few fungal species have exhibited promising LAC and lignin-modifying peroxidase synthesis [8,43–47]. *P. chrysosporium* is one of the most investigated species among a range of pollutants degrading fungi by its extracellular enzymes

including; LiP, MnP, and LAC production processes [6,8,48–50]. Although other WRBs from other taxonomic groups have been assessed, additional research is required to identify potential species and its associated nutritional and culture conditions for effective productivity and increased yield of LMEs. In contrast to fungi, bacterial machinery is comparatively less developed for production of degrading enzymes and proper folding protein which could be able to work in an optimum environment [28]. The catalytic activities of LMEs have been employed to oxidize a wide range of phenolic and non-phenolic pollutants, including; endocrine-disrupting chemicals (EDCs), polycyclic aromatic hydrocarbons (PAHs), polychlorinated dibenzodioxins (PCDDs), polychlorinated biphenyls (PCBs), dibenzofurans, and pharmaceuticals and personal care products (PPCPs), and non-steroidal anti-inflammatory drugs (NSAIDs) [11,51–57]. As concerning their extended application, LMEs can act sufficiently on lignin polymer, which is produced enormously from pulp & paper industry. Conversely, enzymatic biotransformation of kraft lignin has been documented extensively in the last two decades, enumerating the transformed low molecular weight (LMW) compounds after treatment either through enzyme or LME-producing species under controlled conditions [8,58–64]. As a byproduct, an enormous quantity of technical lignin serves as a rich source of aromatic compounds that may be further valorized in the synthesis of value-added compound [65–69]. Much effort has been made to develop novel techniques for extracting and converting lignin into value-added compounds, including lipids, polyhydroxyalkanoates (PHAs), vanillin, and other aromatic monomers with the goal of lowering the total costs of the biorefinery process [10,70–72]. Interestingly, the strong oxidizing capabilities of LMEs have been examined in computer-aided research to better comprehend enzyme-pollutant interaction behavior, which could possibly be incorporated into real-time assay to accelerate biodegradation [73–75]. The aforementioned computer-aided framework for virtual screening of LMEs assisted pollutant catalysis often relies on docking and MD-simulation to estimate binding affinity, chemical bonding, and associated crucial amino acid residues, which is believed to be a recent breakthrough in environmental research [74–80]. Despite the fact that the protein constituents of LMEs are not identical with

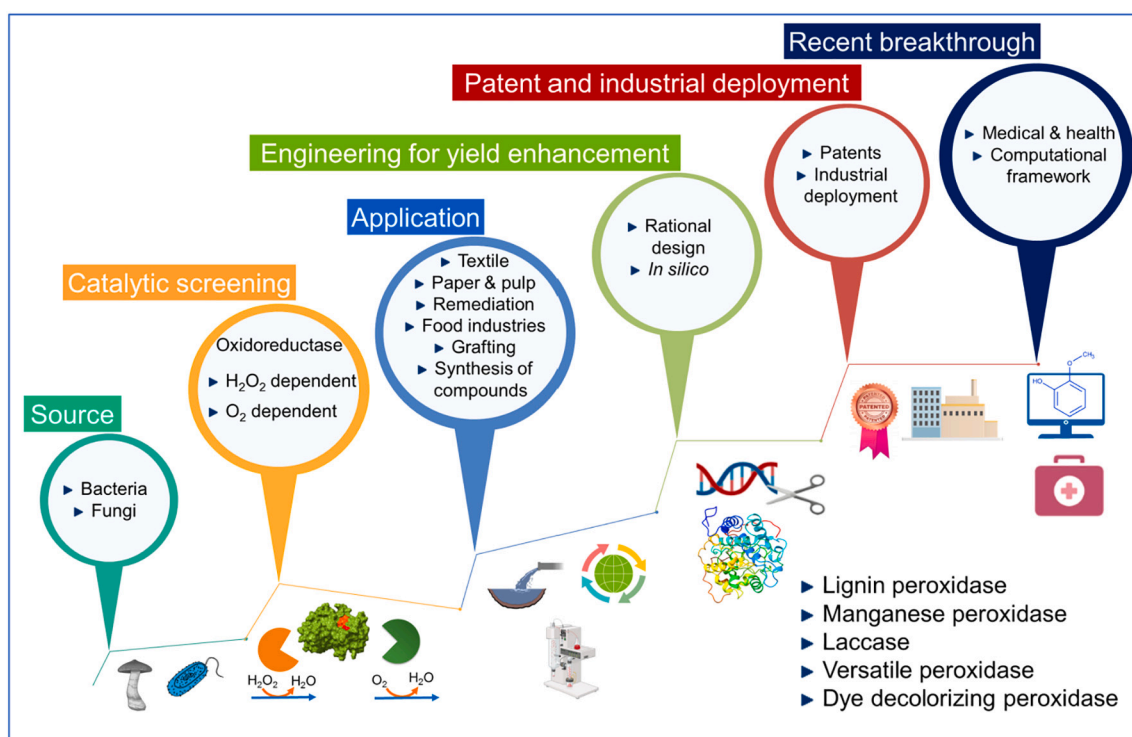


Fig. 1. Highlights of successful milestones in lignin modifying enzymes (LMEs) applications.

respect to helix, sheet, and loop components, therefore pollutant binding mechanism and catalytic activity varies across LMEs members [74,79,80]. Nevertheless, computational finding must be validated utilizing real-time degradation assay to completely understand the biotransformation of parental pollutants, to completely comprehend post degradation toxicity, and fate. In the presented state-of-art, we reviewed the more recent applications of LMEs in biotechnological and industrial research through illustration sketches. Computational aspects and crucial protein features of LMEs have been highlighted. Special emphasis has been furnished on the computational framework, which appears a breakthrough in the last decade as a smart green remediation approach. Furthermore, considerably more significant applications of LMEs in biotechnology, industry, environmental, grafting on materials deployment, computational research, and medical have been outlined with concluding remarks.

## 2. Lignin modifying enzymes (LMEs) producing microbial species

LMEs are extracellular enzymes that are often produced by a few bacterial and fungal species [28,81–84]. Bacterial species including; *Brucella*, *Ochrobactrum*, *Sphingobium*, and *Sphingomonas* genera belonging to the class  $\alpha$ -Proteobacteria have been reviewed for producing enzymes that act on lignin [81]. Likewise, another class  $\gamma$ -Proteobacteria including *Pseudomonas fluorescens* (LiP-producing), *Pseudomonas putida* (DyP, MnP-producing), *Enterobacter lignolyticus* (catalase/peroxidase HPI and DyP-producing) and *Escherichia coli* (LAC-producing) have been reviewed by Janusz et al. [81]. Bacterial delignification is both less effective and slower than fungal delignification, and bacterial lignin breakdown is correspondingly limited [84,85]. Bacteria, on the other hand, are increasingly being recognized for their efforts in this process [86,87]. WRF is one of the most studied fungal species which have an efficient lignin-degrading capability in nature [88]. A few potential fungal species have been documented recently for the secretion or production of LMEs in nature, including; *Phlebia floridensis*, *Trametes versicolor*, *Volvariella volvacea*, *Dichomitus squalens*, *Daedalea flavida*, *Phlebia radiata*, *Trametes villosa*, *Trametes pubescens* and many others [89–91]. Extracellular ligninolytic oxidoreductase enzymes such as LAC and peroxidases, as well as other secreted metabolites, facilitate lignin depolymerization by WRF [88,92]. Fungi need more time in contrast to bacteria, often weeks, to develop and generate extracellular enzymes that break down lignin, as outlined in recently reported research [85,93,94]. Additionally, fungal LMEs work at weak acid to neutral pH (4–7) and diverse temperature ranges (30–55 °C) for the activity of the degradation and decolorization processes [35–37]. Several White-Rot Basidiomycete (WRB) has been examined in recent years to identify viable LMEs, while a few fungal species have exhibited promising LAC and lignin-modifying peroxidase synthesis [8,43–47]. Table 1 highlights the few well-studied microbial species that have been identified as LMEs producers.

## 3. Structural features and catalytic mechanisms of LMEs

LMEs contain unique molecular and chemical functionalities, which can be deployed in a wide variety of diverse applications, in three aspects 1) valorization of lignin, 2) depolymerization of lignin, and 3) oxidative functionalities [1,121–123]. Fig. 2 portrays the schematic oxidative catalytic activity of LMEs on lignin polymer. Molecular and structural aspects are a vital part of LMEs for exploring secondary structure element (SSE), protein topology, and its atomic dynamics against pollutants binding. The protein data bank (<https://www.rcsb.org>) has a repository of the crystal structure information on LMEs, which contains plenty of entries from different origins [124,125]. As described above, the LMEs group encompasses enzymes such as LiP, LAC, VP, MnP, and DyP [28,126]. All members except the LAC are hydrogen peroxidase (H<sub>2</sub>O<sub>2</sub>)-dependent oxidoreductase, in catalytic

**Table 1**

Reported microbial species for lignin modifying enzymes production. Species has been explained with its substrate utilization for enzyme production.

LMEs producing microbial species	Produced enzyme	Substrate	Reference
<b>Fungal species</b>			
<i>Bjerkandera adusta</i> TBB-03	LAC	Acetaminophen, bisphenol A, sulfamethoxazole	[95]
<i>Coprinus comatus</i>	LAC	Reactive Dark Blue KR, Reactive orange 1, Reactive Red X-3B, Congo red	[96]
<i>Coriopsis gallica</i>	LAC	Reactive Black 5, Bismark Brown R	[97]
<i>Coriopsis polyzona</i>	LAC	Acid Red 299, Direct Blue 1, Direct Red 28, Disperse Red 1, Disperse Yellow 3, Reactive Black 5, Reactive Red 4, Reactive Yellow 81	[98]
<i>Cyathus bulleri</i>	LAC	Reactive Orange 1	[99]
<i>Daedalea dickinsii</i>	LAC, MnP	Disperse violet-63 Disperse orange-30	[100]
<i>Datronia caperata</i>	LAC	Orange II	[101]
<i>Ganoderma applanatum</i>	LAC, LiP, MnP	Celecoxib, diclofenac and ibuprofen	[57]
<i>Grammothete fuligo</i>	LiP, MnP, LAC	Organosolv, Kraft lignin	[102]
<i>Hexagonia hirta</i>	LAC	Orange II	[101]
<i>Hypholoma fasciculare</i>	LAC	Acid Red 299, Direct Blue 1, Direct Red 28, Disperse Red 1, Disperse Yellow 3, Reactive Black 5, Reactive Red 4, Reactive Yellow 81	[98]
<i>Irpex lacteus</i>	MnP	Remazol Brilliant Violet 5R, Direct Red 5B	[103]
<i>Ischnoderma resinotum</i>	LAC, MnP	Reactive Black 5, Reactive Red 22, Reactive Yellow 15	[104]
<i>Laetiporus sulphureus</i>	LAC, LiP, MnP	Celecoxib, diclofenac and ibuprofen	[57]
<i>Lenzites elegans</i>	LAC	Congo Red	[105]
<i>Oudemansiella canarii</i>	LAC	Congo red	[106]
<i>Peniophora</i> sp. CBMAI 1063	LAC	Syringaldazine (SGD)	[107]
<i>Trametes trogii</i>	LiP, MnP	ABTS (2,2'-azino-din(3-ethylbenzothiazoline-6-sulphuric acid)	[108]
<i>Phanerochaete sordida</i>	LAC	Reactive Black 5	[109]
<i>Pleurotus djamor</i>	LAC, MnP, LiP	Naproxen and ketoprofen	[110]
<i>Pleurotus eryngii</i>	VP	Enhanced pH and thermal stability	[111]
<i>Pleurotus ostreatus</i>	MnP, VP	Orange II, Reactive Black 5 (RB5), (DMP), Phenol Red, Orange II, Reactive Black 5 (RB5), (DMP), Phenol Red	[112,113]
<i>Pleurotus sajor-caju</i>	LAC, MnP	Congo Red, Orange G Reactive Black 5 (RB5) decolorization	[114,115]
<i>Polyporus tenuiculus</i>	LAC	Orange II	[101]
<i>Emmia latemarginata</i>	VP, MnP, LiP	Remazol dye, disperse-dye, indigo dye	[116]
<i>Mucor circinelloides</i>	VP, MnP, LiP	Remazol dye, disperse-dye, indigo dye	[116]
<i>Stropharia rugosoannulata</i>	LAC	Acid Red 299, Direct Blue 1, Direct Red 28, Disperse Red 1, Disperse Yellow 3, Reactive Black 5, Reactive Red 4, Reactive Yellow 81	[98]
<i>Trametes versicolor</i>	LAC	Diclofenac, flufenamic-acid, bisphenol A, Acid Red 299, Direct Blue 1, Direct Red 28, Disperse Red 1, Disperse Yellow 3, Reactive Black 5, Reactive Red 4, Reactive Yellow 81	[98,117]
<b>Bacterial species</b>			

(continued on next page)

Table 1 (continued)

LMEs producing microbial species	Produced enzyme	Substrate	Reference
<i>Agrobacterium</i> sp.	DyP	Lignin	[118]
<i>Comamonas testosteroni</i>	DyP	Lignin	[118]
<i>Klebsiella pneumoniae</i> NX-1	LiP, LAC, MnP	Kraft lignin	[119]
<i>Ochrobactrum tritici</i> NX-1	LiP, LAC, MnP	Kraft lignin	[119]
<i>Pseudomonas fluorescens</i>	DyP	Lignin	[118]
<i>Pseudomonas putida</i> NX-1	LiP, LAC, MnP	Kraft lignin	[119]
<i>Streptomyces ipomoeae</i> CECT 3341	LAC	Lignin	[120]

mechanism [127]. The mechanism of LMEs substrate oxidization has been described in the subsequent sections. The three-dimensional protein structure of all LMEs members has shown in Fig. 3; the related SSE (i.e., alpha helices, and beta-sheets, turn, coils) has reported in Table 2. Nevertheless, the composition of amino acids and other elements are relatively variable in all members of LMEs and feasible to act as oxidoreductase on a wide range of substrates [74,75,79,80].

### 3.1. Lignin peroxidase (LiP)

LiP (EC 1.11.1.14) is a high redox potential (1.4 V) extraordinarily active heme (Fe)-containing enzyme that acts on a wide range of lignin, lignin-based compounds, non-phenolic, and phenolic model compounds of lignin [2,128–130]. The crystal structure of LiP has been deposited concerning fungal sequence (PDBs: 1LGA, 1LLP, 1QPA, 1B80, 1B85, 6ISS, 3Q3U), however, bacterial crystal structure is very limited or lack in compare to fungal species in the PDB database so far [131–134]. For

an example crystal structure of LiP from *Trametes cervina* (PDB: 3Q3U) has 338 amino acids count in a single chain, while heme (Fe) co-factor is the metallic component of this protein which is competent to perform  $H_2O_2$ -dependent oxidative cleavage of numerous bonds in the substrates i.e. lignin [135,136]. The calculated molecular weight of the LiP is roughly 35 kDa [137]. Alpha helix (Hh), extended strand (Ee), beta turn (Tt), and random coil (Cc) make up 35.80 %, 7.69 %, 4.14 %, and 52.37 %, respectively, of the SSE of LiP 3Q3U (Table 2). A few similar enzymes including LigD from *Sphingobium* sp. SYK-6 bacteria (PDB: 4Y9D) are known for acting on lignin by  $\beta$ -aryl ether ( $\beta$ -ether) cleavage has been reported to functioning those of LiP. Such LigD comprised 305 amino acids count in a single chain, and has a calculated molecular weight of about 32.3 kDa [137,138]. In contrast to fungal LiP, it comprises alpha helix (Hh), extended strand (Ee), beta turn (Tt), and random coil (Cc) making up 49.18 %, 13.77 %, 8.52 %, and 28.52 %, respectively, of the SSE. LiP has been extensively documented for cleaving the C-C and C-O-C (ether bond) linkage present in lignin, and oxidizing benzyl alcohols to aldehydes [22]. Moreover, hydroxylation, quinone formation, and cleaving of the aromatic ring are also reported [139]. A considerable variety of non-phenolic lignin model compounds, particularly  $\beta$ -O-4 linkage type compounds, are readily oxidized by LiP [28,75,84]. The LiP oxidation mechanism involves the formation of radical cations with one-electron oxidation, which leads to the cleavage of the side chain, demethylation, intra-molecular incorporation, and molecular rearrangements on reacting substrate [140]. A detailed LiP chemical catalysis/depolymerization scheme has portrayed in Fig. 4. Bacterial-derived LiP has been reported for efficient KL degradation experimentally but lacks protein crystal structure availability at repository databases [28,59,141,142]. WRF species are substantially more efficient than bacteria on lignin depolymerization, xenobiotic degradation, and decolorization of dyes [4,42,143–146]. Numerous studies have reported recombinant/engineered LiP/DyP, which has demonstrated significantly enhanced catalytic capabilities in an unstable environment [147–150]. A structural representation of LiP and heme (Fe) binding

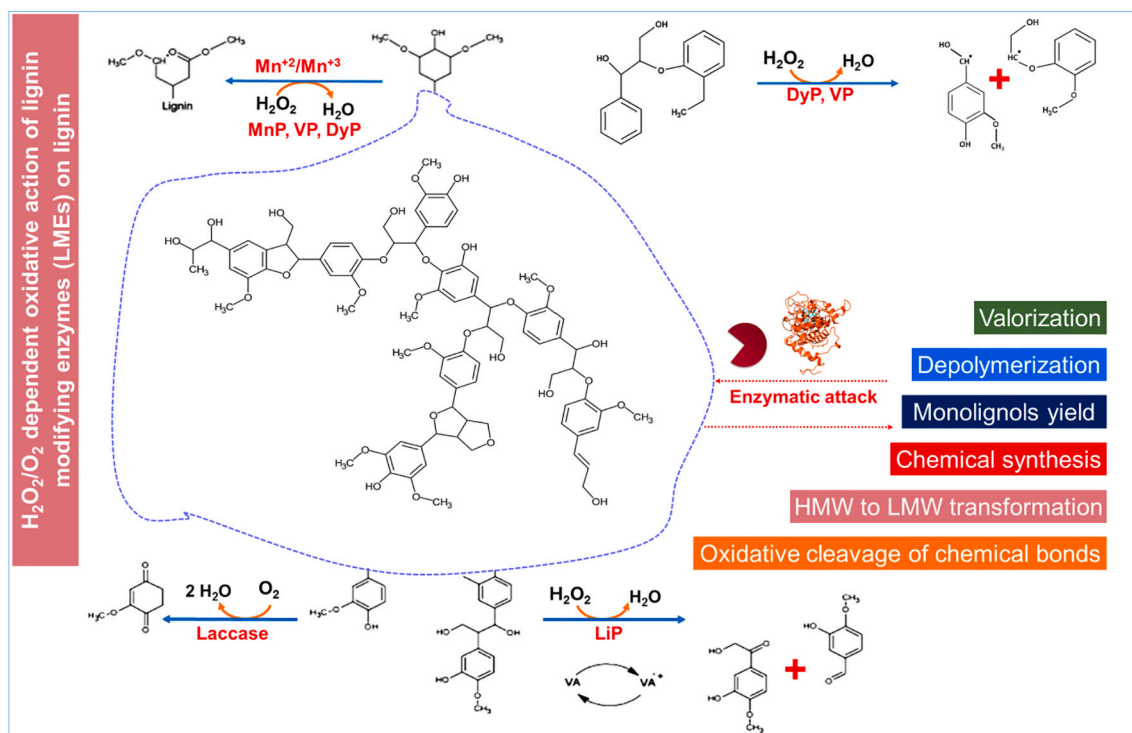
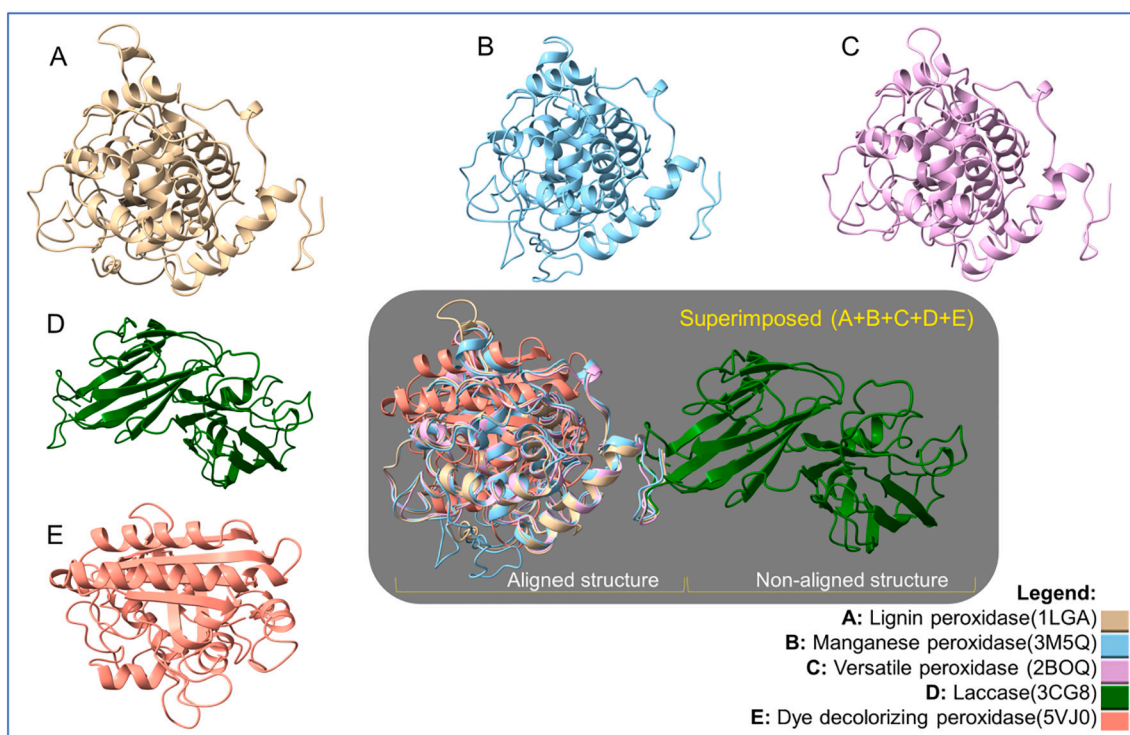


Fig. 2. Chemical catalytic mechanism of lignin modifying enzymes (LMEs). Peroxidases (LiP, MnP, VP, and DyP) share  $H_2O_2$  dependent catalytic mechanism, whilst LAC utilizes molecular  $O_2$  instead of  $H_2O_2$ . The most often used substrate of LMEs is lignin reported in the center, while the right panel highlights the chemical functionalities.



**Fig. 3.** Three-dimensional rendered structure of lignin modifying enzymes (LMEs). Protein structures have been re-rendered from respective PDB files. A: exhibited cartoon rendered of LiP; B: exhibited cartoon rendered of MnP; C: exhibited cartoon rendered of VP; D: exhibited cartoon rendered of LAC; E: exhibited cartoon rendered of DyP. Middle panel exhibited the superimposed rendered structures, which conclude that LAC has different structural element and exclude from clusters within RMSD of 2 Å.

**Table 2**

Secondary structural elements (SSE), of LMEs. Each member has listed for different SSE in percentage contribution. LAC can be observed for relatively less helix element, whilst higher in beta component.

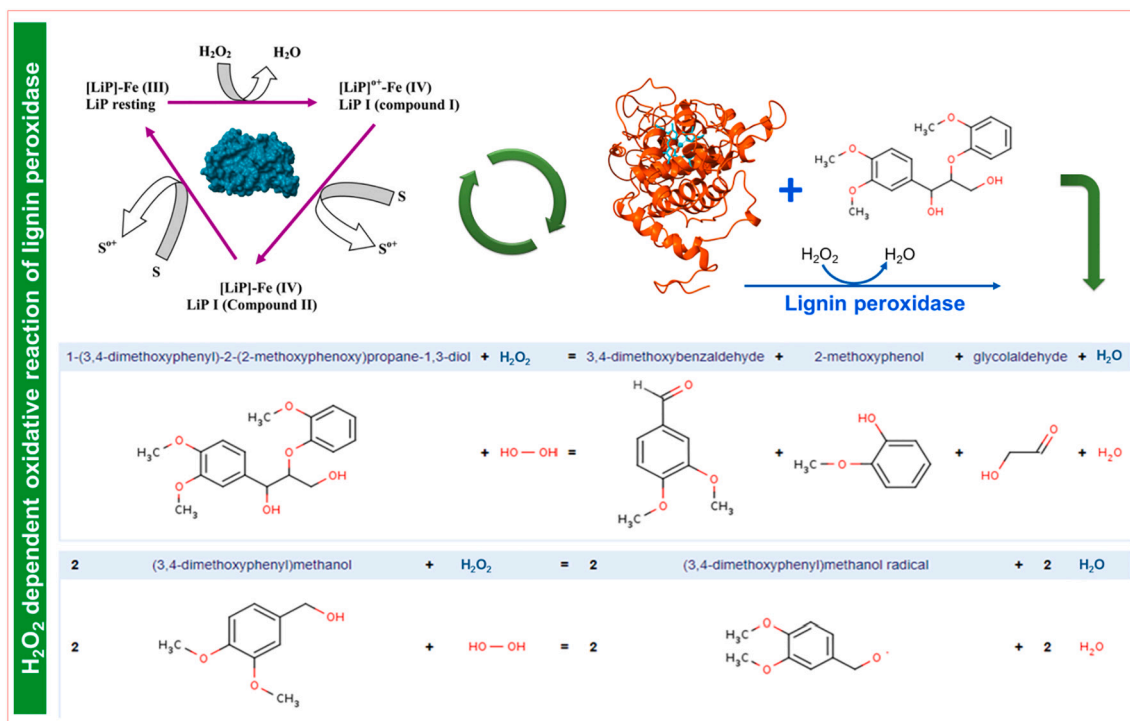
PDB ID of enzyme	Origin	Amino acid residues count	Secondary structure elements(SSE)				Reference
			Alpha helix (Hh)	Extended strand (Ee)	Beta turn (Tt)	Random coil (Cc)	
LiP (3Q3U)	Fungal	338	35.80 %	7.69 %	4.14 %	52.37 %	[222,223]
MnP(3M5Q)	Fungal	357	30.81 %	9.52 %	4.48 %	55.18 %	[190,223]
VP (2BOQ)	Fungal	331	35.95 %	6.65 %	4.53 %	52.87 %	[220,223]
LAC (3CG8)	Bacterial	343	14.29 %	25.66 %	7.29 %	52.77 %	[223,224]
DyP(5VJ0)	Bacterial	318	34.91 %	18.55 %	3.77 %	42.77 %	[223,225]

residues, and active site has portrayed in Fig. 5.

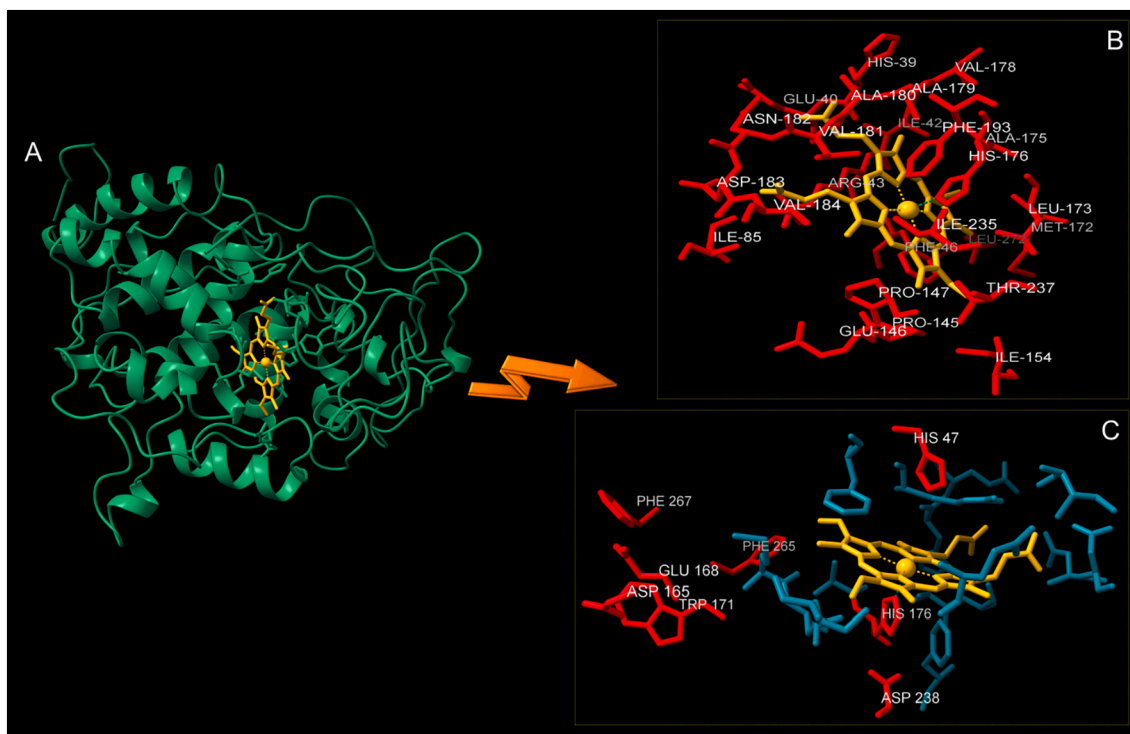
### 3.2. Laccase (LAC)

LACs (EC 1.10.3.2) are multicopper-containing oxidoreductases (MCOs) that oxidize diverse phenolic, non-phenolic, and lignin model compounds utilizing molecular oxygen ( $O_2$ ) as an electron acceptor, resulting in water generated as a by-product through a chemical reaction (Fig. 2) [151–155]. Unlike other member of LMEs, LAC utilizes  $O_2$ , and highly specialized in phenolic substrate catalysis, with and without mediators [156,157]. Since the discovery of the first laccase in *Rhus vernicifera* from the Japanese lacquer tree *Toxicodendron vernicifluum* in 1883, LACs have been reported in a variety of higher plant species [15]. Besides the plants, it also been identified in bacterial, and insects species [158–162]. Most of these enzymes, however, have been isolated from fungi, which are common LAC producers, and deployed into use for degrading lignin amid the decomposition of wood [15,161,163,164]. Adequate protein crystal structures of LACs from various origin species are available in the Protein Databank (<https://www.rcsb.org>), enabling structural and molecular investigations in research of interest [125]. The structural features of different LAC with different origins including;

plants (*Zea mays*: 6KLG), fungi (*Trametes versicolor*: 1KYA), bacteria (*Streptomyces coelicolor*: 3CG8), and insects (*Drosophila melanogaster*: NP\_651441.1) have explained in subsequent figs [134,165–168]. The SSE of LAC (3CG8), consisting of alpha helix (Hh), extended strand (Ee), beta turn (Tt), and random coil (Cc) make up 14.29 %, 25.66 %, 7.29 %, and 52.77 %, respectively [136]. In contrast to LiP, LAC has the lowest helix elements and higher beta component in its protein architect. The crystal structure of *Streptomyces coelicolor* origin LAC (PDB: 3CG8) with a calculated molecular weight of around 36.8 kDa comprises 343 residues in three chains (A, B, and C) [125,137,169,170]. Likewise comparable crystal structure of LAC of fungal origin (*Trametes versicolor*), PDB: 1KYA comprises 499 residues in four chains (A, B, C, and D) [171]. Crystal structure of LAC from plant origin (*Zea mays*), PDB: 6KLG comprises 550 residues in a single chain [172]. The protein structural architecture of LACs from a different source of origin has explained in Fig. 6. Multi copper binding site of LAC has portrayed in Fig. 7. LACs are divided into two categories based on their redox potential: low- (from 0.4 to 0.5 V) and high-redox potential (from 0.5 to 0.8 V) enzymes [159,173,174]. In contrast to the great prevalence of high-redox-potential LAC in fungi, low-redox-potential enzymes are found in bacteria, plants, and insects [159,175]. Gram-positive and gram-negative



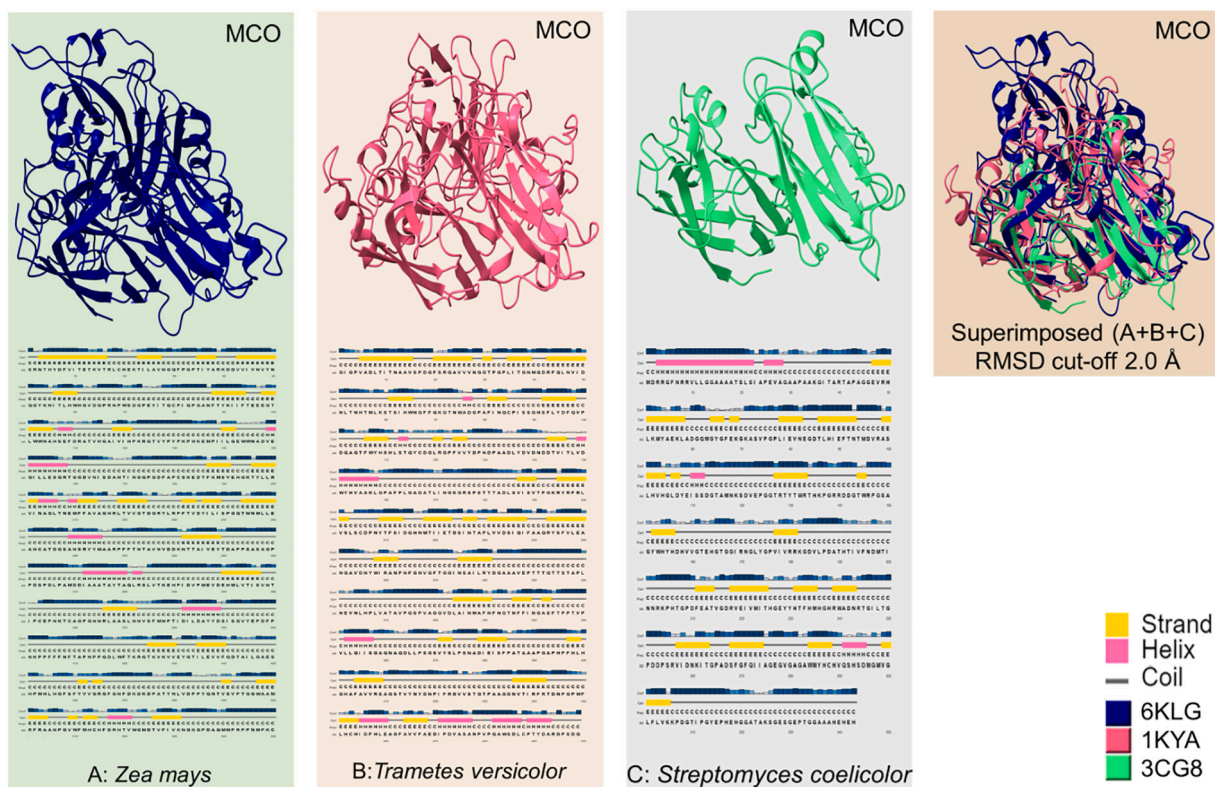
**Fig. 4.** Catalytic attributes of lignin peroxidase acting on lignin polymer. Left upper panel depicts the basic catalytic scheme, whilst right mechanism of dimer compound catalytic activity by LiP. Bottom panel depicts the detailed illustration of lignin dimer ( $\beta$ -O-4) model compound cleavage.



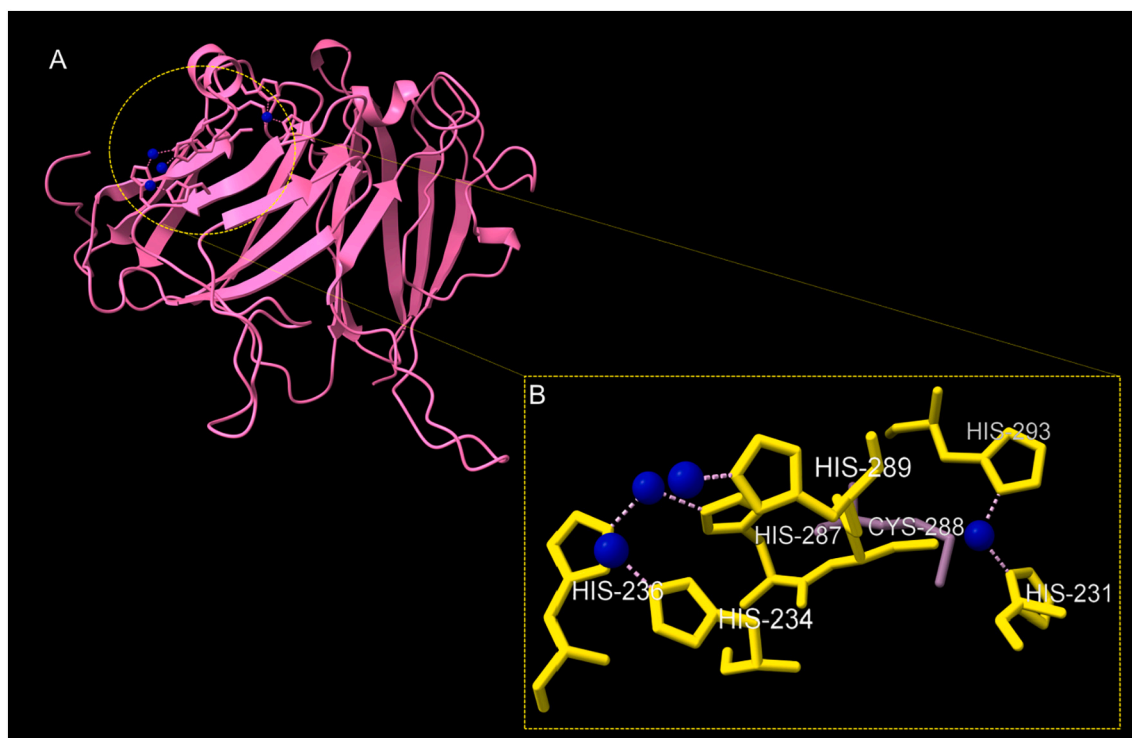
**Fig. 5.** Structural analysis of lignin peroxidase (PDB ID: 1LGA). A: depicts the rendered image of LiP with heme (Fe) in center. B: predicted interactions of amino acid residues corresponding to heme atom. C: depicts the active site of LiP for substrate binding. However, active site is substrate dependent, might be change with different substrate binding. A, and B has been re-rendered with respective PDB.

bacteria from soil and water that are a part of the phyla  $\alpha$ - and  $\gamma$ -proteobacteria, *Firmicutes*, *Cyanobacteria*, *Aquificae*, and *Deinococcus-Thermus*, as well as bacteria from the class Archaea, has been recognized for producing prokaryotic LAC [176]. The redox potential of bacterial and

fungal LACs have changed throughout evolution, this lead to broadening the variety of complex substrates that may be oxidized. It has been noted that LAC from *Ganoderma lucidum* is more stable at low temperatures (30 °C) and an acidic pH of 3.0 than other bacterial LACs [177].



**Fig. 6.** Protein structure of Laccases-multi-copper-oxidase (MCO). A: highlights the protein structure of LAC from *Zea mays* in the upper panel, whilst protein secondary structure element topology in the bottom panel. B: highlights the protein structure of LAC from *Trametes versicolor* in the upper panel, whilst protein secondary structure element topology in the bottom panel. C: highlights the protein structure of LAC from *Streptomyces coelicolor* in the upper panel, whilst protein secondary structure element topology in the bottom panel. A clear structural differences can be visualized in the structural topology of LAC from different members with a cut-off RMSD in 2.0 Å, which exhibited similar enzymes has different structural architect which are not aligned in superimposed rendered (upper right panel).



**Fig. 7.** Protein structure of Laccases-multi-copper-oxidase (MCO). A: Highlights the protein structure of LAC, has been rendered from respective PDB IDs. B: Highlights the predicted copper binding site residues corresponding to copper atoms.

Notwithstanding, only a few bacterial and fungal origin LACs have been widely exploited in biotechnological applications and environmental studies including food industries, biocatalysis, pharma, cosmetics, agrochemicals, bioremediation, sensor, nanobiotechnology and biomedicine [178–180]. In biotechnology, they are characterized as a “Green Tool” or “Green Catalysts” for a wide array of biotechnological applications [181–183]. With the remarkable potential in oxidative catalysis, LAC has gained interest in various biotechnical applications in previous decades [151]. Catalytic reaction triggers the cleavage in a unique way, i.e., C-C cleavage, oxidation, and alkyl-aryl cleavage. LACs have broad substrate specificity; hence can catalyze phenolic and nonphenolic compounds efficiently [163,184]. The detailed catalytic mechanism of LAC has portrayed in Fig. 8.

### 3.3. Manganese peroxidase (MnP)

MnP (EC 1.11.1.13) is a heme (Fe), and manganese (Mn)-containing extracellular enzyme frequently synthesized by many bacterial and fungal species (WRF) [24,108,185]. Several MnP-producing microorganisms (bacterial, fungal, mainly WRF) have been well documented that acts on lignin, including lignin degradation, lignin valorization, and bioremediation [24,126,186–188]. MnP has also been used in dyes, phenol and pharmaceutical products degradation [24,189]. The crystal structure of MnP (PDB: 3M5Q) from *Phanerothontia chrysosporium* has a single chain with 357 residues and has a calculated molecular weight about of 37.45 kDa [137,190,191]. The SSE of MnP (3M5Q) has reported in Table 2. The substrate action mechanism of MnP is an oxidation-reduction biochemical process that follows the ping-pong mechanism and encompasses second-order kinetics in enzymology. MnP works in the presence of H<sub>2</sub>O<sub>2</sub> or biologically produced peroxidase. It employs one cofactor heme (Fe) and Ca<sup>2+</sup> is essential for carrying out the catalysis. The chemical reaction of MnP involves the one-electron oxidation of Mn (II) to Mn (III), which in turn oxidizes a large number of phenolic substrates. The chemical reaction mechanism has been explained in Fig. 2. More precisely, in the initial step, H<sub>2</sub>O<sub>2</sub> enters the active site of MnP, where it binds to Fe (III) ion in the heme cofactor to

form a heme peroxidase complex (Fig. 2). After that, two electrons are transferred from Fe<sup>3+</sup> to peroxidase, which breaks the oxygen-peroxidase bond to form H<sub>2</sub>O and Fe (IV) oxo-porphyrin radical complex [192]. MnP catalyzes Mn (II) to Mn (III) peroxide-dependent oxidation. The Mn (III) is released in complex oxalate from the enzyme such that the oxalate-Mn (III) compound is thus used as a diffused redox mediator that oxidizes lignin [193].

### 3.4. Dye decolorizing peroxidase (DyP)

DyP (EC 1.11.1.19) is a recently identified family of heme (Fe)-containing peroxidase, which has received significant attention owing to its ability to dye decolorization, degradation of lignin, and other compounds [23,121,194–196]. This enzyme is also hydrogen peroxide (H<sub>2</sub>O<sub>2</sub>) dependent, as an electron acceptor to catalyze the oxidation of a wide range of substrates, including lignin, dyes, and other organic compounds [197]. DyP has been expressed in bacterial species and a few other eukaryotic organisms (fungal species) [126,198–200]. DyPs have been categorized into different groups depending on their sequence features as A, B, C, and D types [200]. Several DyPs are included in the protein databank (<https://www.rcsb.org>), under the keyword “lignin peroxidase” based on lignin as the active substrate. A considerable number of DyP-producing bacteria have been reported in recent years, even though it is consistent with the investigation that verified DyP genes are commonly prevalent in bacterial genomes [51,194,201]. The crystal structure of DyP from *Enterobacter lignolyticus* (PDB: 5VJ0) has 318 residues dispersed in four chains (A, B, C, and D) with a molecular weight of about 35 kDa [125,137,194]. The SSE of DyP (5VJ0) has reported in Table 2. Few bacterial species, including *Bacillus subtilis* KCTC2023, have been studied for their ability to degrade lignin and a few dyes, including reactive blue 19 and reactive black 5 [202]. DyP enzymes cleave the C-C linkage or bond without the presence of any oxidative mediator [203,204]. DyP enzymes have long been exploited in the decolorization of dyes and the remediation of lignin and phenolic compounds [29,31,147,205]. DyPs-assisted applications in dyes removal from wastewater, lignin content removal, oxidation of lignin

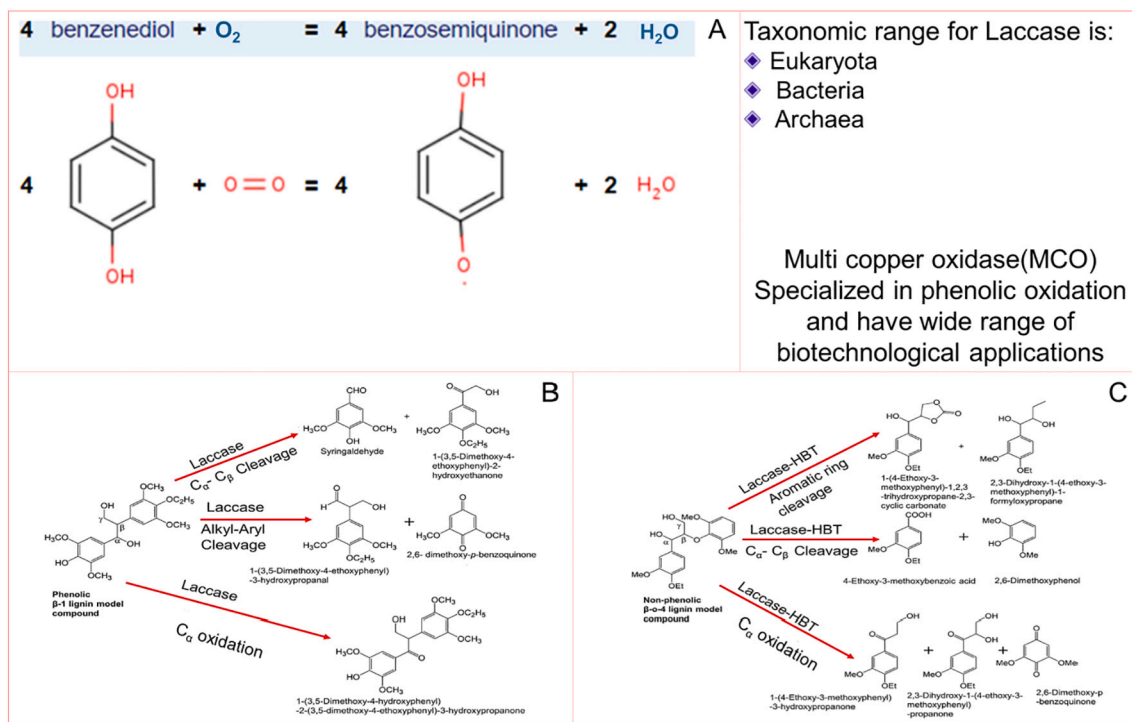


Fig. 8. Laccase (multi-copper-oxidase (MCO)) catalytic mechanism. A: Highlights the basic oxidative mechanism of LAC. B: Illustrative scheme of phenolic lignin model compound ( $\beta$ -1) catalysis. C: Illustrative scheme of non-phenolic lignin model compound ( $\beta$ -O-4) catalysis.



model compounds, phenolic compounds oxidation, and other biotechnological applications have increased in published literature due to their excellent catalytic activity in recent years [122,196,201,206–210]. Many recent investigations have revealed the role of DyP in the catalysis of  $\beta$ -O-4-unit lignin model compounds as well [75,118,147,211]. Fig. 2 represents the catalytic mechanism of DyPs similar to heme peroxidase (LiP).

### 3.5. Versatile peroxidase (VP)

VP (EC 1.11.1.16) is a heme (Fe)-containing oxidoreductase with a hybrid molecular architecture that combines many oxidation-active regions, allowing high redox catalytic capacity for a wide range of substrates [212–214]. Given its strong redox potential, VP has been a robust biocatalyst for widespread industrial applications [215]. A range of bacteria and fungi have been documented for VP production and their active application in bioremediation of environmental contaminants [42,216–219]. From structural perspective, the crystal structure of VP from *Pleurotus eryngii* (PDB: 2BOQ) consists of 331 residues distributed in a single chain, with a molecular weight of 34.6 kDa [137,220,221]. The SSE of VP (2BOQ) has reported in Table 2. The VP catalysis mechanism is similar to that of oxidoreductase family enzymes (LiP, MnP). At the active sites of VP,  $H_2O_2$  and cofactor initiate catalysis. VP's catalytic versatility is advantageous for low and high-redox-potential aromatic substrates in  $Mn^{3+}$ -mediated and Mn-independent reactions. Phenolic compounds, synthetic dyes, and lignin may all be oxidized using  $H_2O_2$ -dependent VP, which employs ferric heme center to catalyze the oxidation of two molecules of reducing substrate with concomitant oxidation of  $Mn^{2+}$  to  $Mn^{3+}$  [213]. The specific catalytic mechanism of VP has been portrayed in Fig. 2.

## 4. Industrial and biotechnological applications of LMEs

Despite the remarkable deployment in diverse array of application, LMEs has been commercialized by different vendors and available in

pure form [226–229]. LMEs have been reported in wide range of applications including; bioremediation, biofuel, biobleaching in pulp & paper industries, textile, biosensor, bio-based compound synthesis, cosmetics, pharmaceuticals, wastewaters treatment and so on [180,230,231]. LMEs serve as vital biocatalysts that conduct eco-friendly degradation/catalysis of an array of environmental pollutants (e.g., EDCs, PCBs, NSAIDs, lignin, phenolics, chlorinated phenol, etc.) from environmental matrices owing to their broad redox potential range [5,232]. Over the last several decades, there has been a surge in research on LAC's potential as biocatalysts to replace traditional chemical processes in the textile, pulp & paper, and pharmaceutical sectors [180]. The cosmetic, paint, and furniture industries are just an application that might benefit from these enzymes [180]. Furthermore, LACs have a role in the lignocellulose-based feedstock used in the manufacturing of bioethanol [180]. The investigation of LAC and potential applications in industrial and biotechnology is a rapidly expanding research area [180]. LiP has also been shown to have a possible biotransformation functioning in the industrial-scale synthesis of vanillin from different substrates [233]. Despite the various features like, relatively low production cost, comparably high yield (LAC, MnP), short cultivation, high thermostability, and improved enzymatic activities have rendered LMEs mass deployment in industrial and biotechnology applications. Nevertheless, issues with cost, stability, and recycling/reuse rates of LAC's limit their use in biotechnological applications [234]. A few detailed applications of LMEs have been outlined in subsequent sections. A schematic illustration of LME-mediated deployment in industrial and biotechnological applications has highlighted in Fig. 9.

### 4.1. Pulp and paper industries: remediation, and bio-bleaching

Lignin causes a barrier in pulp for making paper, therefore its removal is a key step in cellulosic biomass's industrial applications, including the achievement of bioethanol and the manufacture of products such as paper and other products made with cellulose [235]. The growing production rates of paper and pulp industries, including by-

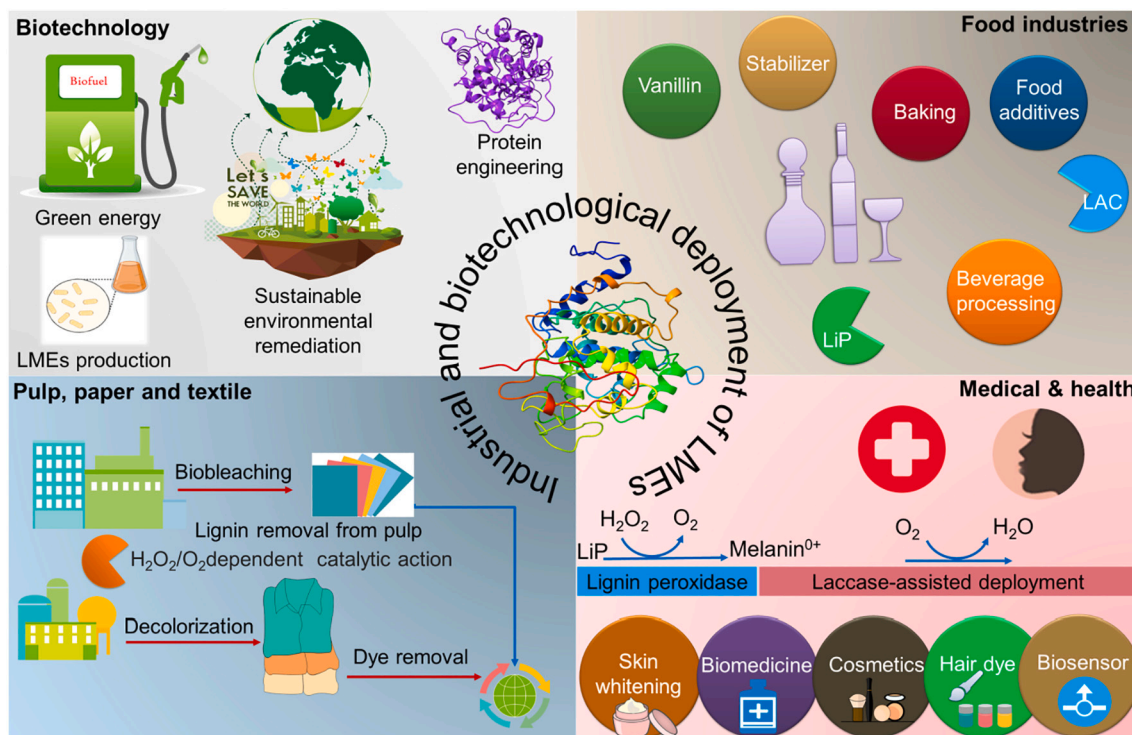


Fig. 9. Schematic illustration representation of industrial and biotechnological deployment of lignin modifying enzymes (LMEs). The different panel depicts the different applications of LMEs in biotechnology, pulp, paper, textile industry, food industry, and medical & health sector.

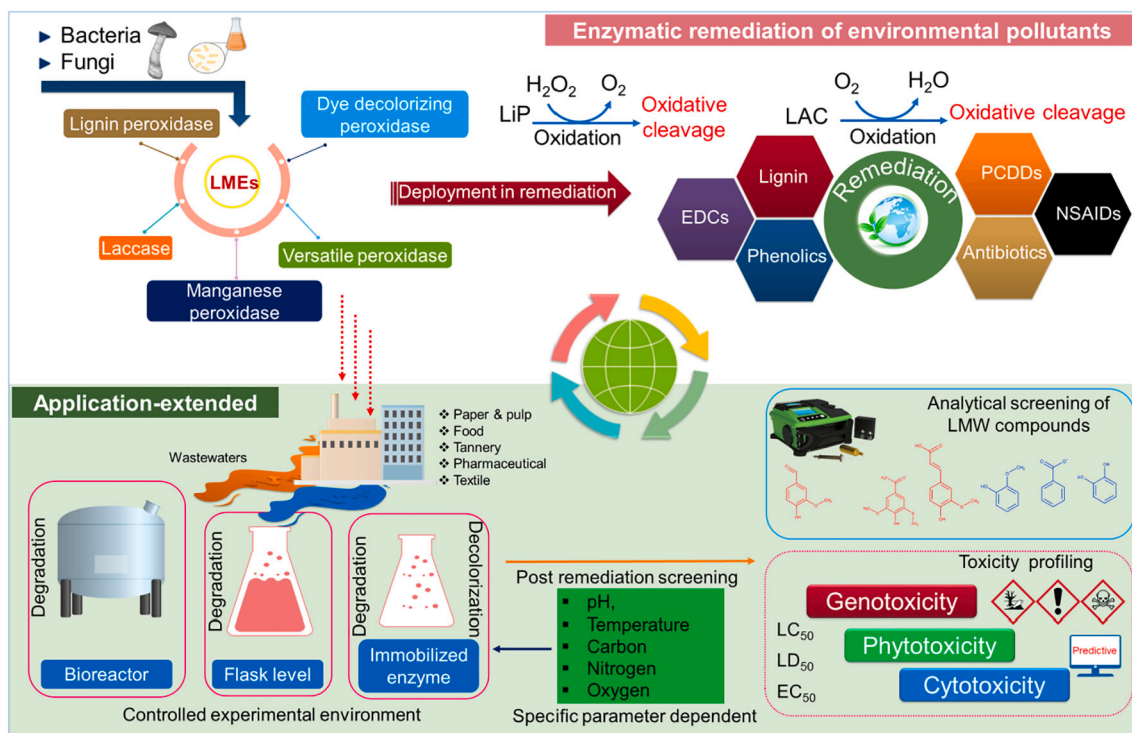
products (Black Liquor) and wastewater, have caused significant environmental issues, and are accountable for environmental repercussions. However, efforts have been made to mitigate the hazardous effects of effluent from paper industries via laboratory research using a variety of toxicological tests [59,236]. Unfortunately, industry-scale studies have been less performed in this direction. Assays on cytotoxicity, chromosomal aberration, and seed germination suggest a significant toxic impact induced by wastewaters of pulp & paper industries [59,236]. Lignin degradation from wastewaters of pulp & paper industries has been performed by exploiting both bacterial and fungal species. A VP has recently been reported in *Pleurotus* and *Bjerkandera* species, the former genus including species being able to degrade lignin selectively [237,238]. Fungal LMEs have an excellent catalytic ability for a wide range of substrates rather than bacteria. The breaking of C $\alpha$ -C $\beta$  and C4-ether linkages results in the release of aromatic aldehydes, one of the principal products identified during enzymatic depolymerization of lignin [235]. LACs have no redox potential enough to directly attack on lignin. When redox mediators are present, they may break down such molecules as well as other recalcitrant compounds efficiently [61,235].

LMEs have been used as bio-bleaching agents commercially in the paper industry with quite an impressive efficiency [239–241]. For the white texturing of paper, lignin must be eliminated from the pulp. The current process is based on Kraft (chemical) pulping method, and sodium hydroxide (NaOH) and sodium sulfide (Na<sub>2</sub>S) treatment at high temperature and pressure to selectively breakdown lignin while retaining the polysaccharide fibers [1,242–244]. Although poor pulp yield is achieved with the krafting process, even targeted polymer is degraded during treatment [242]. To oppose and cope with such challenges, bio-pulping offers an eco-friendly breakdown of lignin from pulp employing either ligninolytic enzymes or ligninolytic microorganisms [245–248]. Bio-pulping softens the wood while increasing paper quality and lowering energy and chemical consumption [247,248]. Numerous fungal species, including WRF, have been extensively investigated for their potential use in the bio-pulping of wood [248–250]. However, the

yield and cost of enzyme production remain a key challenge in this sector. Orozco Colonia et al. demonstrated significant hemicellulose (26.27 %) and lignin (36.80 %) removal with high pulp yield (74.36 %) using LiP and xylanase synthesized by *Aspergillus* sp. during bio-pulping and biobleaching [148,251]. In comparison to current technologies, the use of LiP may assist in reducing the manufacturing costs of expensive chemicals while also allowing for more eco-friendly treatments of pulp in papermaking steps. However, the biobleaching sector is in a research and development stage along with other uses of enzymes in the pulp and paper industry; the greatest current usage includes sustainable biobleaching for hardwood and soft pulp [252]. A schematic illustration of LMEs in remediation and decolorization of industrial wastewaters, and associated toxicity reduction has explained in Fig. 10.

#### 4.2. Textile effluent treatment and dye decolorization

About 10,000 different dyes and pigments, mostly used in the dye and printing industries, are being manufactured annually worldwide [253]. The chemical contents of synthetic dyes are frequently diversified and classified into distinct groups e.g., azo, triphenylmethane, and heterocyclic/polymeric compounds. Synthetic dyes account for a significant portion of the textile sector and are a major source of global environmental pollution [254,255]. Even though extensive use of various dyes in textile industries meets the need for quality or finished items, the manufacturing process produces a large volume of colorful, toxic wastewater [255]. In fact, during the dyeing processes, 10–15 % of dyes escape into wastewater. Many of these dyes are extremely resistant to light, temperature, and microbial degradation, making these into recalcitrant and toxic chemicals [253]. A variety of successful wastewater treatment solutions (activated carbon, constructed wetland, ion exchange, etc.) have been developed for textile water treatment and reuse [256–260]. Unfortunately, the constructed wetland seems to have weak color reduction efficiency while utilizing a comparatively large, occupied area for implementation. Activated carbon appears to have a



**Fig. 10.** Application of lignin modifying enzymes (LMEs) in industrial wastewater treatment, decolorization, and toxicity reduction. The upper panel explains the LMEs producers; i.e. fungi, bacteria, and their assisted oxidative deployment in pollutant remediation from water matrices. While the bottom panel describes the extended application exploitation under controlled environment, and post treatment toxicity profile in a variety of choice for assuming the toxic risk assessment.

reasonable removal rate for water-soluble dyes. However, it cannot adsorb suspended solids (SS) or insoluble dyes from wastewaters. These strategies are effective but costly and create a massive amount of sludge that can further lead to secondary pollution problems [261,262]. As a result, microbial or LME-based treatment offers an environmentally friendly alternative for textile water treatment by lowering dye concentration in water matrices and allowing for further water reusability. Recently, a few studies have reported the efficient decolorization of textile effluent employing microbial LMEs [253,263,264]. This has been recognized for years and has proven to be the most robust bioremediation method to decolorize and metabolize dyes components [23,265]. Parshetti et al. reported that purified LiP from *Kocuria rosea* MTCC 1532 was able to decolorize a total of 11 different dyes; azo, triphenylmethane, heterocyclic, polymeric, metallic complexes with a neutral pH (7.0) [266]. Another study reported decolorization and biodegradation of Disperse Red 3B exploiting a consortium made up of the fungus *Aspergillus* sp. XJ-2 and the microalgae *Chlorella sorokiniana* XJK; this system exhibited stronger efficiency in terms of decolorization (98.09 %) and COD removal [267]. The most recent azo dye RB5 biodecolorization has been shown by Fernandes et al. [268]. In their study crude enzyme from *Pleurotus sajor-caju* was employed as the source of LMEs, with the employment of the “artificial neural network” based study in combination with the genetic algorithm and the response surface methodology (RSM) for the prediction of RB5 decolorization [268]. Subsequently, when LMEs producing fungus was grown in pulp wash, the fungi showed higher LAC activity, whereas when immobilized, a higher MnP activity was achieved, but both allowed decolorizing of the dye in about 89.4 % and 75 %, respectively [268].

#### 4.3. Lignin biodegradation, depolymerization, and valorization

Pulp & paper mill effluent has a negative effect on aquatic life because of its chemical make-up and the fact that it is often quite dark or black, probably due to high amounts of lignin [269–274]. Lignin obtained from the kraft pulping process has a complex structure with a prevalence of C-C, and aryl ether linkages, which are recalcitrant and non-hydrolyzable bonds, turn lignin into challenging for chemically and biologically breakdown [2,275,276]. In previous decades massive studies have been performed to *ex-situ* breakdown of kraft lignin, employing ligninolytic producing bacteria and fungi i.e. *P. chrysosporium* and *Serratia liquefaciens* [59,277–285]. Indeed, lignin biodegradation has been studied to evaluate the enzymatic capability and biotransformation of lignin compounds [286–289]. A certain set of motif/linkages are present in lignin polymer (e.g.,  $\beta$ -O-4,  $\alpha$ -O-4, 5-5,  $\beta$ - $\beta$ , 4-O-5,  $\beta$ -5,  $\beta$ -1), which are the target points to be act by ligninolytic/LMEs [2,290]. LMEs cleave such motifs using an oxidative mechanism as above discussed to yield the simplest structures and monomers. Lignin catalytic depolymerized into monomers and oligomers from lignin polymer have explained in Table 3. In recent years, a broad array of fungi including WRF, derived enzymes has been described for the transformation of xenobiotic chemicals, lignin, and organic pollutants [6,40,42,291–294]. Enzymatic or LME-producing organisms for lignin depolymerization/valorization have been recognized at a large scale for manufacturing value-added compounds for various applications including second-generation biofuels [12,70,141,281,295–297]. Aromatic monolignols, PHAs, lipids, and bio-oils have been identified as key lignin-depolymerized products [298,299]. Valorization of lignin into value-added compounds by the ligninolytic activity and associated microbial species has explained in Table 4. Even LACs have indeed been documented for biotransformation and bioremediation of water contaminants [161]. A detailed illustration of LMEs in lignin depolymerization and valorization has highlighted in Fig. 11.

#### 4.4. Food industry

The employment of LMEs provides a vast potential for upgrading

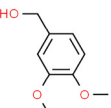
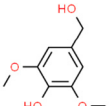
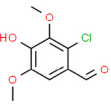
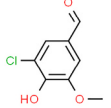
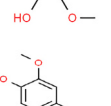
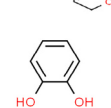
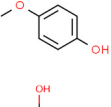
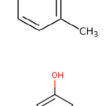
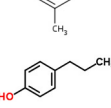
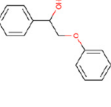
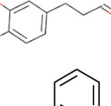
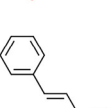

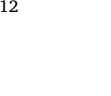
current manufacturing processes and establishing new technologies to obtain high-value products. Enzymes offer cleaner and more efficient production processes and support the sustainable concept. Among LMEs, LACs are exceptionally versatile biocatalysts and have been adopted for 1) removal of phenolic compounds and by-products from food industries, 2) use in food application as enhancers, preservatives, etc. Dedicated applications of LAC and peroxidase in food industries are listed in Table 5. LACs have been employed at an industrial scale in baking, fruit, wine stabilization, etc. [334]. Sugar beet pectin gelation and the stabilization of wine and beer, fruit-juice processing are just a few of the many potential applications of LAC in food industries [335,336]. The phenolic compounds contained in wine are accountable for its color, flavor, and overall taste [180,336]. Therefore, LAC may be used to selectively removal of polyphenols, that retain the wine's organoleptic properties [337]. Beer storage life may be improved by using LAC to remove O<sub>2</sub> at the completion of the brewing process [180]. To prevent the formation of off-flavor precursors, LAC eliminates O<sub>2</sub> that might otherwise react with fatty acids, amino acids, proteins, and alcohols [180]. LAC has also been studied in baking to enhance dough machinability and end-product softness, as well as in teas and oil-containing products for enhancing flavor and color [180]. Furthermore, the gelling effects of LAC in blackcurrant juice, luncheon meat, and milk with added sugar beet pectin have been investigated [180]. A recent research evaluated the influence of LAC and LMS on stirred milk yoghurt in a process that mimicked industrial manufacturing [338]. LAC treatment resulted in protein breakdown at the molecular level, while the inclusion of the natural redox mediator vanillin triggered the formation of higher molecular weight oligomers [180]. LAC convert O<sub>2</sub> to H<sub>2</sub>O, and the biosensor then measures the amount of oxygen utilized during analyte oxidation. LAC-based sensors have been extensively utilized in the food sector for determining polyphenols in fruit juices, wine, and teas, as well as to quantify fungal contamination in grape musts [180]. Another LMEs, LiP, has received little attention in the food industry. Vanillin is a remarkable use of LiP in the food industry. Lignin-based artificial vanilla is more often employed as a flavoring agent in foods, drinks, and medicines [339]. Biocatalytic transformations of aromatic substrates such as lignin and guaiacol into vanillin with LiP activity has been reported by exploiting purified LiP and LiP-producing WRF *P. chrysosporium* [148,340,341]. Saikia et al. documented magnetically separable co-immobilized LAC and VP-based framework for transforming lignocellulosic biomass into vanillin [342]. Subsequently depolymerization of lignin by free and immobilized LAC showed a vanillin yield of 24.8 % and 23 %, respectively, at pH 4.0 in 6 h at 30 °C against a vanillin yield of 20 % and 21.7 % by the free and immobilized VP, respectively, at pH 5.0 and 50 °C [342].

### 5. Emerging contaminants degradation and wastewater treatment

Despite their environmentally friendly nature, LMEs-members have been applied in the treatment of wastewater from a variety of sources, including food industries, tanneries, textiles, pulp & paper, and distilleries [159,178]. Several tons of pharmaceutical chemicals flow through the wastewater treatment system each year [346,347]. Emerging contaminants (EC) are contaminants identified at trace concentrations of worldwide drinking water sources at which human and environmental health hazards are not yet recognized or little known. Pharmaceutical compounds and their accompanying contamination are reported to be EC as they have been identified as a possible source of wastewater treatment plants (WWTPs), which have detrimental effects on surface water when utilized for drinking water if water bodies receive wastewater. NSAIDs are one of the most recalcitrant EC in the aquatic environment [348]. The occurrence of NSAIDs drug products in the environment and wastewater has been regarded as an EC, which is of great concern in the scientific community because of the toxic impact [348]. NSAIDs are one of the principal causes of wastewater pollution;

**Table 3**

A catalytic depolymerized product of lignin into monomers and oligomers model compounds has listed with chemical and molecular formula.

Lignin derived compound	Type	Structure	Molecular formula	Molecular weight (g/mol)	Reference
Veratryl alcohol	Monomer		C <sub>9</sub> H <sub>12</sub> O <sub>3</sub>	168.19	[300]
Syringyl alcohol	Monomer		C <sub>9</sub> H <sub>12</sub> O <sub>4</sub>	184.19	[301]
2-Chlorosyringaldehyde	Monomer		C <sub>9</sub> H <sub>9</sub> ClO <sub>4</sub>	216.62	[302,303]
5-Chlorovanillin	Monomer		C <sub>8</sub> H <sub>7</sub> ClO <sub>3</sub>	186.59	[304]
Sinapyl alcohol	Monomer		C <sub>11</sub> H <sub>14</sub> O <sub>4</sub>	210.23	[305]
Guaiacol	Monomer		C <sub>7</sub> H <sub>8</sub> O <sub>2</sub>	124.14	[306]
Coniferyl alcohol	Monomer		C <sub>10</sub> H <sub>12</sub> O <sub>3</sub>	180.20	[305]
Catechol	Monomer		C <sub>6</sub> H <sub>6</sub> O <sub>2</sub>	110.11	[300]
4-Methoxyphenol	Monomer		C <sub>7</sub> H <sub>8</sub> O <sub>2</sub>	124.14	[307]
m-Cresol	Monomer		C <sub>7</sub> H <sub>8</sub> O	108.14	[306]
p-Cresol	Monomer		C <sub>7</sub> H <sub>8</sub> O	108.14	[306]
4-Propylphenol	Monomer		C <sub>9</sub> H <sub>12</sub> O	136.19	[308]
2-Phenoxy-1-phenylethanol	Dimer		C <sub>14</sub> H <sub>14</sub> O <sub>2</sub>	214.26	[309]
4-Propylguaiacol	Monomer		C <sub>10</sub> H <sub>14</sub> O <sub>2</sub>	166.22	[309]
Anisole	Monomer		C <sub>7</sub> H <sub>8</sub> O	108.14	[309]
Cinnamyl alcohol	Monomer		C <sub>9</sub> H <sub>10</sub> O	134.17	[309]

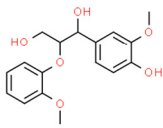
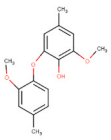
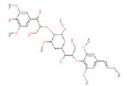
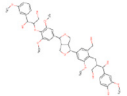





(continued on next page)

Table 3 (continued)

Lignin derived compound	Type	Structure	Molecular formula	Molecular weight (g/mol)	Reference
Vanillyl alcohol	Monomer		C <sub>8</sub> H <sub>10</sub> O <sub>3</sub>	154.16	[309]
Vanillin	Monomer		C <sub>8</sub> H <sub>8</sub> O <sub>3</sub>	152.15	[309]
Veratrol	Monomer		C <sub>8</sub> H <sub>10</sub> O <sub>2</sub>	138.16	[309]
Eugenol	Monomer		C <sub>10</sub> H <sub>12</sub> O <sub>2</sub>	164.20	[309]
Phenol	Monomer		C <sub>6</sub> H <sub>5</sub> OH	94.11	[309]
2-Propylphenol	Monomer		C <sub>9</sub> H <sub>12</sub> O	136.19	[309]
4-Methylanisole	Monomer		C <sub>8</sub> H <sub>10</sub> O	122.16	[309]
Phenyl-phenyl-ether	Dimer		C <sub>14</sub> H <sub>14</sub> O	198.26	[309]
4-Hydroxybenzoic acid	Monomer		C <sub>7</sub> H <sub>6</sub> O <sub>3</sub>	138.12	[310]
Diphenyl ether	Dimer		C <sub>12</sub> H <sub>10</sub> O	170.21	[309]
Benzyl phenyl ether	Dimer		C <sub>13</sub> H <sub>12</sub> O	184.23	[309]
4-Phenoxyphenol	Dimer		C <sub>12</sub> H <sub>10</sub> O <sub>2</sub>	186.21	[309]
Methyl gallate	Monomer		C <sub>8</sub> H <sub>8</sub> O <sub>5</sub>	184.15	[311]
Benzoate	Monomer		C <sub>7</sub> H <sub>5</sub> O <sub>2</sub> <sup>-</sup>	121.11	[312,313]
Ferulate	Monomer		C <sub>10</sub> H <sub>9</sub> O <sub>3</sub> [O <sup>-</sup> ]	193.18	[313]
Ferulic acid	Monomer		C <sub>10</sub> H <sub>10</sub> O <sub>4</sub>	194.18	[314,315]
Acetosyringone	Monomer		C <sub>10</sub> H <sub>12</sub> O <sub>4</sub>	196.20	[316]
Vanillyl alcohol	Monomer		C <sub>8</sub> H <sub>10</sub> O <sub>3</sub>	154.16	[317,318]

(continued on next page)

Table 3 (continued)

Lignin derived compound	Type	Structure	Molecular formula	Molecular weight (g/mol)	Reference
Dimer guaiacylglycerol-beta-guaiacyl ether	Dimer		C <sub>17</sub> H <sub>20</sub> O <sub>6</sub>	320.3	[318,319]
Dimer (guaiacyl 4-O-5 guaiacyl)	Dimer		C <sub>16</sub> H <sub>18</sub> O <sub>4</sub>	274.31	[75,320]
Trimer (syringyl β-O-4 syringyl β-O-4 sinapyl alcohol)	Trimer		C <sub>34</sub> H <sub>44</sub> O <sub>13</sub>	660.713	[75,320]
Tetramer ( guaiacyl β-O-4 syringyl β- β syringyl β- O-4 guaiacyl)	Tetramer		C <sub>43</sub> H <sub>52</sub> O <sub>14</sub>	792.875	[74,320]
Dibenzodioxocin	Dimer		C <sub>14</sub> H <sub>10</sub> O <sub>2</sub>	210.228	[300]
Phenoxyphenol	Dimer		C <sub>12</sub> H <sub>10</sub> O <sub>2</sub>	186.21	[300]
Diphenylmethane	Dimer		C <sub>13</sub> H <sub>12</sub>	168.23	[300]
2-Isopropylphenol	Monomer		C <sub>9</sub> H <sub>12</sub> O	136.19	[300]
Dihydrobenzofuran	Monomer		C <sub>8</sub> H <sub>8</sub> O	120.15	[300]

LMEs have been demonstrated efficient capability in the biodegradation of NSAIDs from water matrices [110,349–352]. Cruz-Ornelas et al. demonstrated biodegradation of naproxen (90 %) and ketoprofen (87 %) using WRF *Pleurotus djamor*, during 48 h of incubation [110]. Bankole et al. reported that a combination of three NSAIDs (celecoxib, diclofenac, and ibuprofen) was biodegraded by *Ganoderma applanatum* and *Laetiporus sulphureus* within the incubation period of 72 h [57]. Similarly, Kasonga et al. reported an investigation to eliminate a range of pharmaceutical compounds (carbamazepine (CBZ), diclofenac (DCF) and ibuprofen (IBP)) from wastewater utilizing the ligninolytic fungus *Rhizopus* sp. with LiP, and MnP activity [353]. Apriceno et al. found that NSAID drugs (diclofenac, naproxen, and ketoprofen) were eliminated by employing a laccase-based approach in their recent investigation [348]. García-Morales et al. reported biotransformation of acetaminophen and diclofenac by immobilized laccase adopting titania nanoparticles, which was able to achieve 90 % and 68 %, biotransformation respectively [178]. Although not extensive, an adequate study has shown remarkable ligninolytic efficiency in the elimination of NSAIDs and other pharmaceuticals with an effective breakdown rate [352,354–357]. Potential environmental clean-up applications of LMEs have been listed in Table 6. Since the end of the 1990s, there has been an increase in the number of compounds suspected of interfering with the endocrine system. These compounds like “hormone mimics” impair the endocrine system and trigger unfavorable human, wildlife, and development reproductive and neurological consequences [358–361]. Approximately 800 substances are known or suspected to interfering with hormone receptors. However, only subsets of these compounds have been studied

in experiments capable of detecting overt endocrine effects in intact organisms [362]. Phthalates, PCBs, PAHs, and chemical additives including bisphenol-A, alkylphenol ethoxylates (APEs), pesticides like dichlorodiphenyltrichloroethane (DDT), and other additives are well-reported examples of EDCs [363–366]. Industrial/anthropogenic activities are among the primary sources of EDCs in the environment; as a consequent EDCs reach into the environment and water matrices [367]. EDCs in the environment and water matrices potentially lead to public health risks once exposed to them through various pathways. Therefore, they must be eliminated from the environment for public health safety. Numerous methods exist for removing EDCs; however, oxidoreductase enzyme-mediated removal has received extensive interest for a few decades in eco-friendly mitigation [368–370]. A few fungal species and LME member, including LiP, VP, and LAC has been reviewed for the effective elimination of a wide array of EDCs from water matrices [371–375]. The degradation of various new emerging pollutants by the ligninolytic enzyme-producing WRF has garnered increasing attention [371,376–378]. Several studies have demonstrated the use of LACs for EDCs removal utilizing either independent or subsidiary LAC-aided approaches [368,369,379–386]. Dai et al. demonstrated bisphenol-A elimination by immobilized laccase in electrospun fibrous membranes modified with carbon nanotubes [387]. Zdarta et al. outlined the biodegradation of bisphenol-A and bisphenol-F by *Trametes versicolor* laccase immobilized on *Hippospongia communis* spongin scaffolds [388]. Under optimal conditions, almost 100 % of BPA (pH 5, 30 °C) and BPF (pH 5, 40 °C) was removed from the solution at a concentration of 2 mg/mL [388].

**Table 4**

Valorization of lignin into value-added compounds by the ligninolytic activity and associated microbial species.

Ligninolytic microbes/enzyme	Utilized substrate	Valorized product	Reference
<i>Cupriavidus basilensis</i>	Kraft lignin	PHA	[321,322]
LAC	Lignin	<i>p</i> -Hydroxybenzaldehyde, vanillin	[323]
LAC, VP	Lignin	Vanillin	[324]
LAC	Lignin	Grafting/lignin-based materials	[325]
<i>Pseudomonas putida</i> KT2440-CJ122	<i>p</i> -Coumaric acid	Pyruvate	[326,327]
<i>Pseudomonas putida</i> KT2440-CJ122	<i>p</i> -Coumaric acid	Lactic acid	[326,327]
<i>Pandoraea</i> sp. ISTKB	Kraft lignin	PHA	[328]
<i>Paraburkholderia aromaticivorans</i> AR20-38	Lignin monomer	Vanillic acid	[310]
<i>Phanerochaete chrysosporium</i>	Lignin	Succinic acid	[329]
<i>Pseudomonas putida</i> KT2440	Lignin	<i>cis,cis</i> -Muconic acid	[330]
<i>Pseudomonas putida</i> KT2440	Model lignin-derived compound	<i>cis,cis</i> -Muconate	[331]
<i>Rhodococcus opacus</i> PD630	Lignin	Lipid	[332]
<i>Rhodococcus opacus</i> PD630	Kraft lignin	Lipid	[333]
<i>Rhodococcus opacus</i> PD630	Alkaline-pretreated lignin	Gallate	[311]
<i>Streptomyces</i> sp. S6	Kraft lignin	3-Methyl-butanoic acid, guaiaicol derivatives, and 4,6-dimethyl-dodecane	[141]

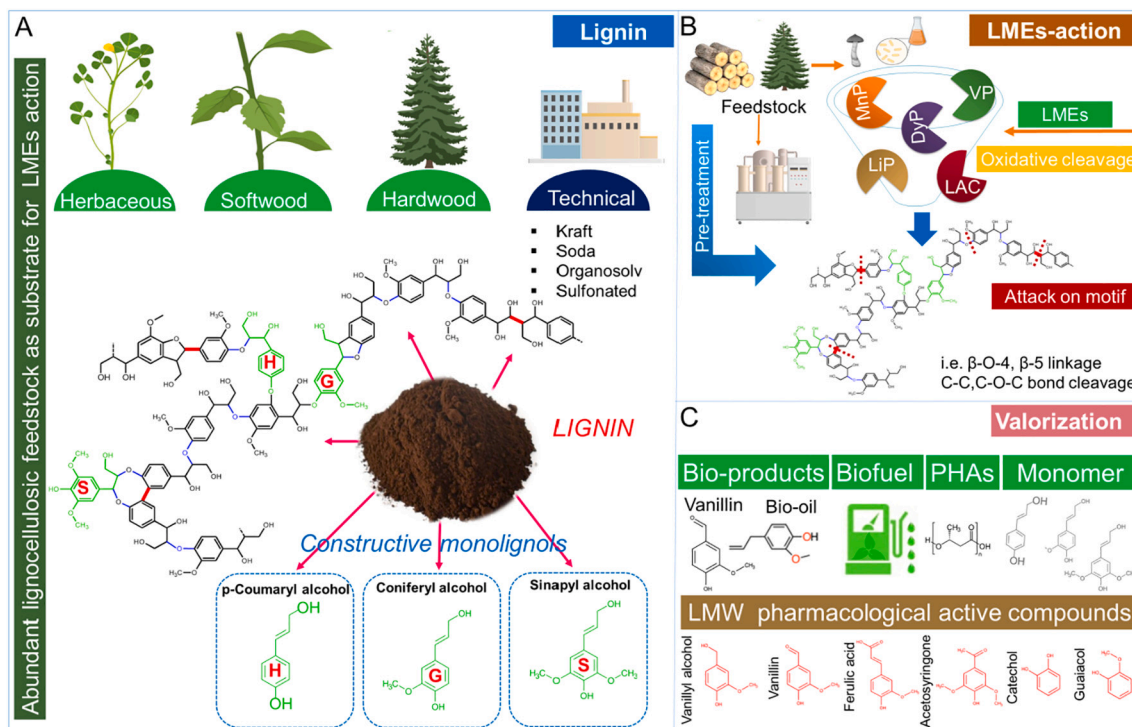
## 6. Medical applications/pharmaceutical and skin whitening agent

Despite their outstanding oxidoreductase characteristics, only a few members of the LME family (i.e. LAC, LiP) has shown significant potential in few medical applications, including skin whitening agents, development of antibiotics, anticancer drugs, antifungal drugs, and sedative as well [179,396]. LiP has been incorporated into skin whitening agents as one of the most recent breakthroughs for melanin decolorization and as an ingredient for treating pigment impairments in humans [397–399]. Many fungal strains have been identified as a source of LiP for melanin decolorization, and potential use in skin lightening applications [398,400,401]. However, the majority of these applications is still in the development phase and has yet to be commercialized [402,403]. In a recent clinical self-controlled study, 31 persons with melasma on both sides of their faces were given an eight-week full-facial therapy [399]. After that, effective skin whitening was documented at

**Table 5**

Addition of lignin-modifying enzymes (LMEs) in food processing steps.

Food applications	Value added properties	Reference
Baking	Increase stability, strength, reduction of stickiness improving machinability of the dough	[343–345]
Beer stabilization	Removal of certain polyphenol	[343,344]
Beverage processing	Enhancing the color appearance	[343,344]
Flavor enhancing	Development of flavor, color, and nutritional quality of food	[345]
Food sensory parameters	Reduction of bitterness	[343,344]
Fruit juice processing	Prevention of the hazes by the interactions between polyphenols and proteins	[343,344]
Sugar beet pectin gelation	Crosslinking the beet pectin, by the mechanism of oxidative coupling of the feruloyl groups	[343–345]
Wine stabilization	Removal of oxygen from finished beer	[343,344]



**Fig. 11.** Catalytic action of lignin modifying enzymes (LMEs) on lignin. A: describes the natural lignin from plants source, and technical lignin along with its types. B: explains the chemical functionalities of LMEs i.e. oxidative cleavage, and attack on lignin motif, to break the chemical bonds and yield lignin derivatives. C: explains the enzymatic-valorization, and value-added compound, fuel, Bio-products, PHAs, and pharmacological monomeric compound yield.

**Table 6**  
Dedicated environmental applications of lignin modifying enzymes in remediation of pharmaceutical compounds (LMEs).

Enzyme	Environmental application	Reference
DyP4 peroxidases	2-Mercaptobenzothiazole (MBT), paracetamol, and furosemide degradation	[26]
LiP	EDCs removal	[375]
VP	Estrone, 17 $\beta$ -estradiol, and 17 $\alpha$ -ethinylestradiol removal	[389]
LAC	Transformation of anti-inflammatory drugs	[117]
LAC	NSAIDs removal	[348]
LAC	Degradation and detoxification of an array of pesticides	[385]
LAC	Emerging contaminants degradation	[368]
LAC	Removal of endocrine disruptors	[390]
LAC	Degradation of Azo dyes	[391]
LAC	Bioremediation of xenobiotic compounds	[392]
LAC	phenol and bisphenol-A removal	[379]
LAC	Xenobiotic degradation	[393]
LAC and peroxidases	Emerging pollutants biocatalysis	[394]
LAC/oxidoreductases	Biocatalytic degradation of pharmaceutically active compounds	[395]
LiP, MnP, LAC	Pharmaceutical and personal care products (PPCPs)	[366]
LiP, MnP, LAC	Endocrine-disrupting chemicals (EDCs)	[366]

specific therapy intervals (0, 7, 28, and 56 days), with no adverse reaction to the skin/facial color palette [399]. The normal skin luminance ( $L^*$ ) values were restored after 7 days of administering the LiP whitening lotion [399]. Another LME member LAC's potential to degrade melanin has also been reported for skin-whitening applications [404].

The use of LACs in medicinal chemistry has shown their potential as an emerging field for the development of antibiotics, anticancer drugs, antifungal medications, sedatives, and hazardous material detoxification [396,405,406]. Likewise, a series of 1,4-naphthoquinone-2,3-bis-sulfides, synthesized with the aid of Novozym® 51003 (*M. thermophila* laccase expressed in *Aspergillus oryzae*) catalysis, showed activity against melanoma (UACC62), prostate (PC3), breast (MCF7), and renal (TK10) cancer cell lines, with some compounds having  $IC_{50}$  values under 10  $\mu$ M [407,408]. Another application has been documented for hair dyeing products in recent years [409]. The principal intermediate (*p*-phenylenediamine, *p*-aminophenol, and its derivatives), the coupler (*m*-phenylenediamines, resorcinol, naphthols, and derivatives), hydrogen peroxide, and ammonia are typically blended to make permanent hair dyes [179]. The oxidant's primary function is to oxidize the primary intermediates; its secondary function, when combined with ammonia, is to lighten the hair's natural color [179,410]. However, these circumstances facilitate adverse interactions with hair protein, resulting in damage to the hair. Therefore, this impact may be lessened by using a different oxidant, such as laccase instead of hydrogen peroxide [179]. It has been reported that alkaline LACs have been used for this purpose as well [409,411].

## 7. Emerging trends in nano-bio catalyst and immobilization

Functional nanostructures have been shown to be suitable supports for the production of enzyme-nanostructure biocatalysts due to their unique physicochemical properties [412]. The development of new nanocatalysts has been linked to significant improved enzyme activity in a variety of applications in recent years [412–414]. Owing to its unique attributes, such as improved enzyme activity and stability under harsh environments, with relatively low concentrations, nano-biocatalysts have sparked intense interest in the bioremediation of wastewater from industrial sectors [366,415]. Recently, Guo et al. reported LiP immobilized from *Pichia methanolica* on  $Fe_3O_4@SiO_2$ @polydopamine (PDA) based nanoparticles for the effective degradation of tetracycline, dibutyl phthalate, 5-chlorophenol, phenol, phenanthrene, fluoranthene,

and benzo(a)pyrene [416]. Singh et al. reported the synthesis of LiP-mediated silver nanoparticles (AgNPs) in *Acinetobacter* sp. for optimal activity of the purified enzyme at pH 2 and 60 °C using *n*-propanol as substrate [417]. Gkantzou et al. reviewed the innovative nanocatalysts for biocatalytic transformation applications, such as the removal of textile dyes from wastewaters employing LAC from *T. versicolor* immobilized on kaolinite [413,418]. In terms of decolorization yield, nano-biocatalysts are often superior to free LAC because to the improved stability brought about by immobilization.

## 8. Laccase-aided grafting

The use of LAC-mediated grafting on lignocelluloses as an environmentally friendly way to covalently alter wood, paper, and cork has received much attention in recent years [419]. This method has also been used to alter fibers having polysaccharide backbones, including cellulose or chitosan, to impart coloration, antibacterial action, or antioxidant activity. Furthermore, particular attention has been dedicated to the grafting of (bio)-molecules onto surfaces and polymers. Grafting is described as a process in polymer chemistry when one or more species of the block are covalently linked to the main chain of a macromolecule as side chains possessing constitutional or configurationally properties that differ from those in the main chain [419]. A few most recent examples of LAC-mediated grafting have explained in subsequent sections. A descriptive schematic illustration of grafting has portrayed in Fig. 12.

### 8.1. Lignocellulose grafting through laccase facilitation

A phenolic radical produced by LAC interacts with another radical or an oxidizable moiety in the process of propagation (termination). A scheme is explained in Fig. 13. However, the free radical is delocalized across the aromatic system, which implies several reactive sites, whether it is produced by phenol in solution or lignin [419]. As a result of coupling such delocalized radicals, various substrate-lignin, substrate-substrate, and even lignin-lignin dimers and subsequent oligomers are generated. The inclusion of phenolic extractives in the lignocellulosic matrix, which may potentially function as LAC substrates, makes it more difficult to comprehend the mechanisms behind these activities. Applying mediators to the system changes the reaction pathway further [420].

### 8.2. Acrylic acid grafting with laccase onto lignin for its recovery from wastewaters

By grafting acrylic acid (AA) to lignosulfonate in the presence of LAC and *tert*-butyl hydroperoxide (*t*-BHP), AA in paint production wastewater could be recovered. Interestingly, low *t*-BHP levels do not inhibit LAC activity but instead increase radical production of lignin, which then triggered AA polymerization [421]. LAC plays a substantial part in the AA grafting to lignin. Laccase/*t*-BHP enables 94 % of AA to be polymerized on lignin, but only 32 % of AA is grafted on lignin under the same conditions without laccase [421].

## 9. Synthesis of bioactive natural compounds and their analogs from LACs

Yet another potential use for LACs is the green and environmentally friendly production of highly valuable organic compounds. I.e. polyphenols, antimicrobial powders, etc. [422–425]. Despite the numerous chemical functionalities including; amination, oxidation, polymerization, dimerization, and cyclization LAC has been focused on the synthesis of numerous chemical compounds [426]. The oxidation of phenolic substrates to the respective quinone structures is a different strategy for using LAC in chemical synthesis, and it has been thoroughly documented [15]. Determining the susceptibility of LAC from fungi,



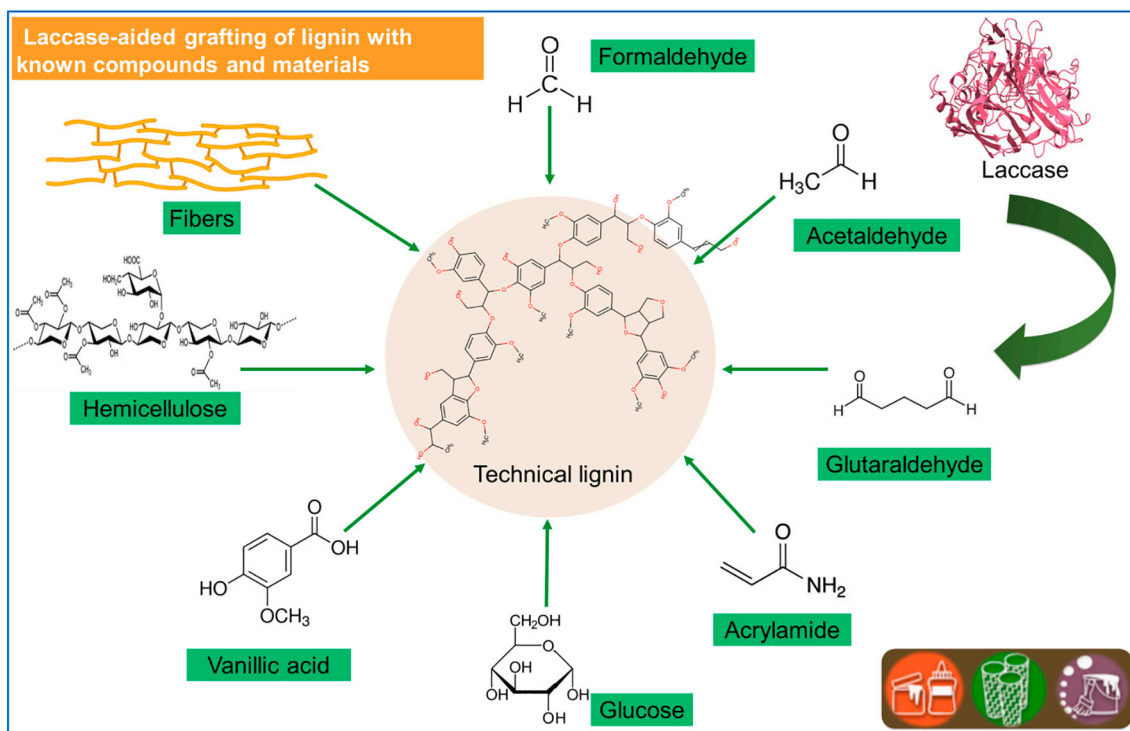


Fig. 12. A basic schematic of laccase-assisted grafting into lignin exploiting different materials.

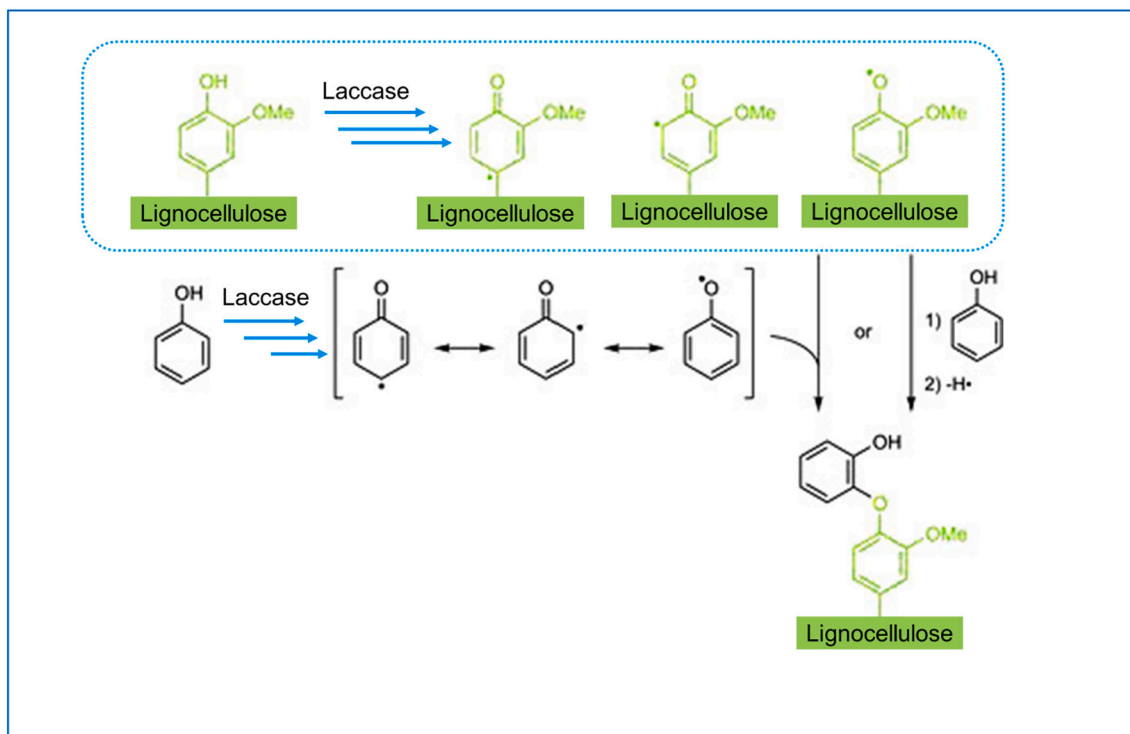


Fig. 13. A basic schematic of laccase-assisted phenol grafting into lignin.

bacteria, and plants to particular substrates and reactions have also been the attention of several investigations. In well-structured publications, comparisons of enzymes from various sources in-depth as well as assessments of several types of LAC from the same source have been reported and discussed [15,427,428]. Most recent, Cardullo et al. reported LACs and peroxidases could be deployed for the synthesis of numerous

organic compounds synthesis i.e. lignans, neolignans, dimeric stilbenoids, dimeric flavonoids, biflavonolignans, biaryl compounds and so on [15,429]. Likewise in another study, Hahn et al. reported LAC mediated ring-closure mechanism for the synthesis of phenothiazines, phenoxazines, and phenazines [422]. LAC's viability may be confirmed through successful lab-to-market products.

## 10. Deployment of LMEs in pre-screening of degradability or theoretical degradation of environmental pollutants

Enzyme kinetics and other parameters are vital in enzyme-substrate reaction, however, lack underlying molecular interactions, and simultaneously biotransformation information [73]. In recent decades, LME-producing microbes have been widely used for lignin degradation, depolymerization, and a variety of biotechnological applications [5,118,430]. For both environmental and various biotechnological uses, they have been labeled “green biocatalysts” owing to their effectiveness, efficiency, and environmental friendliness. Unfortunately, numerous critical pieces of information are unavailable in LME-assisted degradation, including the complete chemical transformation, underlying chemical interplay between enzyme and pollutant, and specific amino acids that interact as active site residues along with the dominant chemical bondings i.e. H-bond,  $\pi$ - $\pi$ , sigma, alkyl, and  $\pi$ - $\pi$  stacking [73]. However, existing shortcomings might be overcome by employing a computational framework in combination with a real-time assay to enhance biodegradation rate. Computational frameworks including docking, MD-simulation, and chemical cleavage prediction have gained the highest influence in predictive biodegradation for revealing the underlying molecular interplay between enzyme and pollutants at atomic level [73,75,431]. In recent years, LMEs has been outlined in computational frameworks for predictive biodegradation that depend on a wide range of aforementioned techniques, including docking and MD-simulation for virtual screening of degradability [73,75]. Even though it is a feasible and robust approach for pre-screening of enzymatic degradation, it has numerous advantages over the inefficient and time-consuming biodegradation process [73]. In contrast to conventional bioremediation, which primarily involves the entire media or the target contaminant of concern, the fate, molecular interactions, and rate of each transformed compound seem to be unidentified, even though the assessment of degradation compounds is based on the analytical probability of GC-MS/MS screening [73,431]. Although computational frameworks have been little documented, they might be considered a green computational tool in the remediation of environmental pollutants for regulatory consideration (Table 7). Over the last several years, several docking and simulation studies have been documented employing lignin and its derivatives as ligand in an effort to elucidate the molecular interaction between LMEs and ligand (pollutants) [77,320,432,433]. Previously, we reported on an *in silico* investigation of bacterial LiP using a set of 12 lignin model compounds to examine

**Table 7**

Exploitation of lignin modifying enzyme in computational degradation of lignin model compounds for unraveling molecular interactions between enzyme-pollutants.

LMEs member	Substrate/model compounds	Application	Reference
LAC	Lignin model compounds	Structural and functional properties of laccase	[77]
LAC	Lignin model compounds	Molecular basis of lignin binding to laccase	[434]
LAC	Lignin model compounds	Lignin degradation	[435]
LAC	Lignin model compounds	Molecular basis of LAC-lignin binding	[436]
LiP	Different lignin model compounds	Molecular basis of LiP-lignin bindings	[75]
LiP	Multimeric lignin model compounds	Theoretical degradation of model compounds	[74]
LMEs	EDCs	Theoretical basis of degradation mechanism	[79]
LMEs	Priority pollutants	Theoretical basis of degradation mechanism	[80]
LAC	Glyphosate, isoproturon, lignin, parathion	Bioremediation potential of laccase	[78]

molecular binding affinity and get a theoretical understanding of the lignin degradation mechanism at the molecular level [75]. Subsequently lowest XP Gscore as  $-8.136$  (kcal/mol) was found for the trimer model compound, with  $\text{Pi-Pi}$  stacking and H-bond type bonding interactions corresponding to LiP [75]. Among all model compounds specific binding residues were observed as TYR, HIP (protonated histidine), PHE, VAL, ASP, THR, LYS and GLN. Furthermore, MD-simulation was deployed for validation of docked complex with a simulation ran of 30 ns [75]. A similar analysis was carried out in another investigation to describe the catalytic activity of LiP against multimeric lignin model compounds to comprehend the theoretical degradation potential of selected LiP [74]. Nevertheless, additional investigation of LMEs against a broad range of hazardous compounds for potential catalytic behavior still remains unexplored. A schematic representation of LME-assisted degradability screening of pollutants has been highlighted in Fig. 14. Future views and the incorporation of computational framework for enhancing bioremediation of various pollutants along with joint efforts including *in silico*, and conventional experiments have been outlined in Fig. 15.

## 11. Concluding remarks and future perspectives

LMEs have made a significant contribution to their prospective abilities in a wide range of industrial and biotechnological applications over the past few decades. Fungi, particularly WRF, have been reported to outcompete bacteria in the biosynthesis of extracellular LMEs enzymes. Extensive application in *ex-situ* environmental studies has conclusively demonstrated their pollutant removal capability, commercial availability, and biotechnological applications. Environmental research has received far more attention than any other application of LMEs. Despite the aforementioned, lower yield and increased enzyme demand remain major challenges in LMEs synthesis by native microbial species. However, protein engineering or recombinant technologies are being developed to tackle such a pillar to meet the growing demand of LMEs for industrial deployment. LiP, MnP, and VP are  $\text{H}_2\text{O}_2$  dependent in the catalysis of phenolic and non-phenolic substrates (lignin), whereas LACs are oxidized phenolic substrates using molecular  $\text{O}_2$ . LAC plays an important role in the food industries as a stabilizer, and destabilizer, pharmaceutical, biosensor, biobleaching, and medical. As opposed to LiP, this possesses a multipurpose oxidoreductase with uses in both the environmental and biotechnological fields. Recent medical advancements and skin whitening agents have been thoroughly documented, proving that LMEs member (LiP, LAC) have exceptional capabilities. The implementation of LMEs in computer-assisted theoretical degradation solves the complicated, recalcitrant pollutant puzzle in a smarter and greener way has brought us even closer to another deployment. LMEs are still expected to provide a number of opportunities for further investigation into previously unknown domains, such as those of:

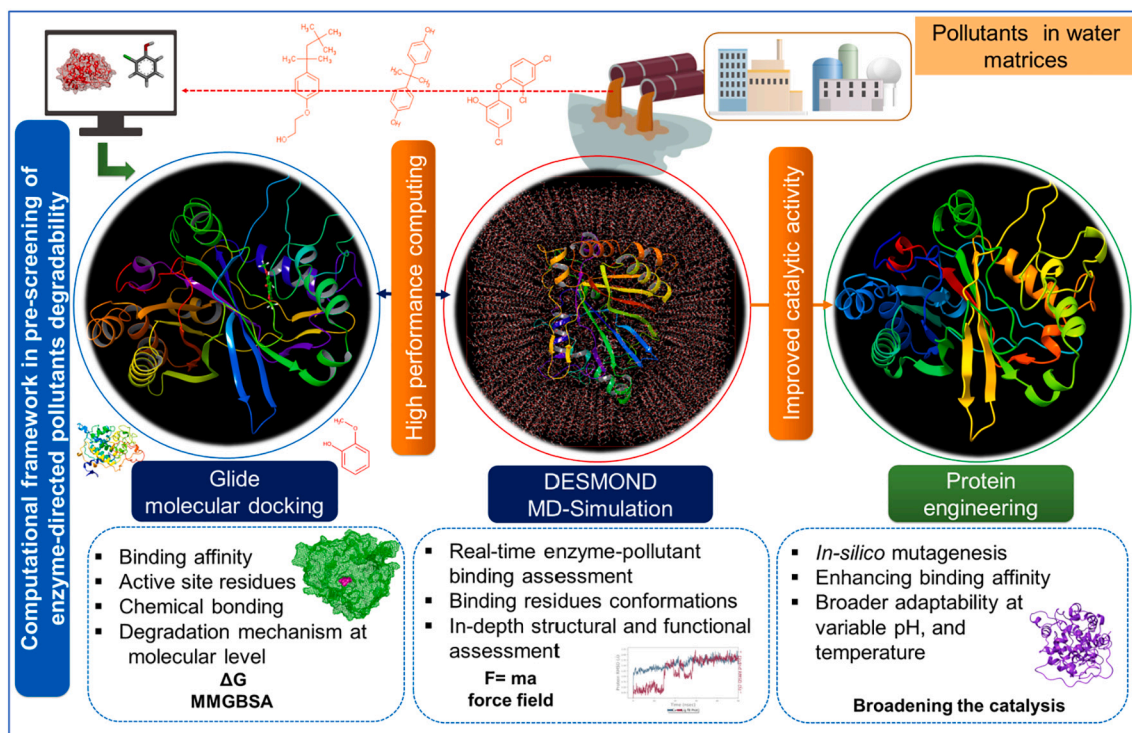
- ✓ Exploration of LMEs-assisted depolymerized compounds screening for drugs properties
- ✓ Protein engineering approach for high yield, pH, and stability at harsh temperature
- ✓ A computational framework for green biodegradation
- ✓ Exploration for extended pharmaceutical and medicinal application
- ✓ Computational exploration of homologous enzyme sequence among other origins e.g., plant species

### CRedit authorship contribution statement

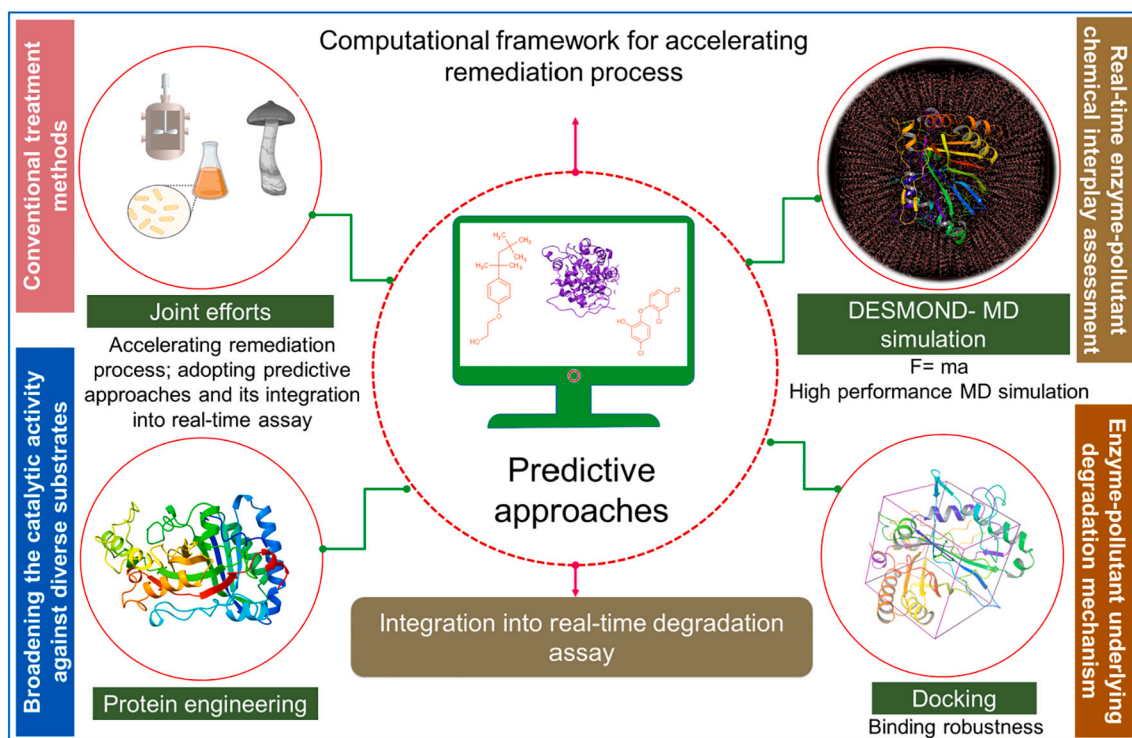
Listed authors have made equal contribution in this manuscript.

### Declaration of competing interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence



**Fig. 14.** A most recent breakthrough in water research. A computational framework has been explained, containing molecular docking, MD-simulation, and protein engineering approaches for sustainable smart remediation of pollutants from water matrices. An array of pollutants from wastewaters could be screened and deployed for docking, and simulation for accessing the degradability and therefore such techniques has designated as pre-screening of pollutants degradability. Different oxidoreductase can be used for target pollutant docking, and simulation, and thus obtained results could be transformed into real-time degradation assay for enhancing the degradation rate. However, the protein engineering approach can be exploited for enhancing the catalytic functionalities under unfavorable environmental conditions.



**Fig. 15.** Future perspective of lignin modifying enzymes (LMEs), for regulatory consideration to access the pre-screening based degradability of pollutants deploying aforesaid member of LMEs. Even though, it would be necessary to confirm the outcomes of the pre-screening methods using a real-time degrading assay.

the work reported in this paper.

## Acknowledgment

AKS would thank the Council of Scientific & Industrial Research (CSIR), India, Academy of Scientific and Innovative Research (AcSIR)-An Institute of National Importance, and CSIR-Indian Institute of Toxicology Research Lucknow, India. The research leading to these results has received funding from the Norwegian Financial Mechanism 2014–2021 under the Project number 2020/37/K/ST8/03805.

## References

- [1] A.K. Singh, M. Bilal, H.M.N. Iqbal, A.S. Meyer, A. Raj, Bioremediation of lignin derivatives and phenolics in wastewater with lignin modifying enzymes: status, opportunities and challenges, *Sci. Total Environ.* 777 (2021), 145988.
- [2] A.K. Singh, M. Bilal, H.M.N. Iqbal, A. Raj, Lignin peroxidase in focus for catalytic elimination of contaminants — a critical review on recent progress and perspectives, *Int. J. Biol. Macromol.* 177 (2021) 58–82.
- [3] Y. Sugano, T. Yoshida, DyP-type peroxidases: recent advances and perspectives, *Int. J. Mol. Sci.* 22 (11) (2021) 5556.
- [4] S. Lee, M. Kang, J.-H. Bae, J.-H. Sohn, B.H. Sung, Bacterial valorization of lignin: strains, enzymes, conversion pathways, biosensors, and perspectives, *Front. Bioeng. Biotechnol.* 7 (2019).
- [5] D. da Silva Vilar, M. Bilal, R.N. Bharagava, A. Kumar, A. Kumar Nadda, G. R. Salazar-Banda, K.I.B. Eguluz, L.F. Romanholo Ferreira, Lignin-modifying enzymes: a green and environmental responsive technology for organic compound degradation, *J. Chem. Technol. Biotechnol.* 97 (2) (2022) 327–342.
- [6] J. Janusz, A. Pawlik, J. Sulej, U. Świdwerska-Burek, A. Jarosz-Wilkolazka, A. Paszczyński, Lignin degradation: microorganisms, enzymes involved, genomes analysis and evolution, *FEMS Microbiol. Rev.* 41 (6) (2017) 941–962.
- [7] L. Pollegioni, F. Tonin, E. Rosini, Lignin-degrading enzymes, *FEBS J.* 282 (7) (2015) 1190–1213.
- [8] V. Elisashvili, E. Kachlishvili, M.D. Asatiani, R. Darlington, K.H. Kucharzyk, Physiological peculiarities of lignin-modifying enzyme production by the White-Rot Basidiomycete *Coriolopsis gallica* strain BCC 142, *Microorganisms* 5 (4) (2017).
- [9] M. Bilal, H.M. Iqbal, Nanoengineered ligninolytic enzymes for sustainable lignocellulose biorefinery, in: *Current Opinion in Green and Sustainable Chemistry*, 2022, 100697.
- [10] L.M.C. Leynaud Kieffer Curran, L.T.M. Pham, K.L. Sale, B.A. Simmons, Review of advances in the development of laccases for the valorization of lignin to enable the production of lignocellulosic biofuels and bioproducts, *Biotechnol. Adv.* 54 (2022), 107809.
- [11] M.R. Mäkelä, K.S. Hildén, J. Kuuskeri, Fungal lignin-modifying peroxidases and H<sub>2</sub>O<sub>2</sub>-producing enzymes, in: Ó. Zaragoza, A. Casadevall (Eds.), *Encyclopedia of Mycology*, Elsevier, Oxford, 2021, pp. 247–259.
- [12] M. Bilal, S.A. Qamar, V. Yadav, H. Cheng, M. Khan, S.F. Adil, M.J. Taherzadeh, H.M.N. Iqbal, Exploring the potential of ligninolytic armory for lignin valorization – a way forward for sustainable and cleaner production, *J. Clean. Prod.* 326 (2021), 129420.
- [13] D.M. Mate, M. Alcalde, Laccase engineering: from rational design to directed evolution, *Biotechnol. Adv.* 33 (1) (2015) 25–40.
- [14] C. Romero-Guido, A. Baez, E. Torres, Dioxigen activation by laccases: green chemistry for fine chemical synthesis, *Catalysts* 8 (6) (2018).
- [15] N. Cardullo, V. Muccilli, C. Tringali, Laccase-mediated synthesis of bioactive natural products and their analogues, *RSC Chem. Biol.* 3 (6) (2022) 614–647.
- [16] E. Contreras, R. Flores, A. Gutiérrez, D. Cerro, L.A. Sepúlveda, Agro-industrial wastes revalorization as feedstock: production of lignin-modifying enzymes extracts by solid-state fermentation using white rot fungi, *Prep. Biochem. Biotechnol.* (2022) 1–12.
- [17] W. Poirier, J.-P. Bouchara, S. Giraud, Lignin-modifying enzymes in *Scedosporium* species, *J. Fungi* (2023).
- [18] A. Iram, A. Berenjian, A. Demirci, A review on the utilization of lignin as a fermentation substrate to produce lignin-modifying enzymes and other value-added products, *Molecules* (2021).
- [19] R. Berni, E. Piasecki, S. Legay, J.-F. Hausman, K.S. Siddiqui, G. Cai, G. Guerriero, Identification of the laccase-like multicopper oxidase gene family of sweet cherry (*Prunus avium* L.) and expression analysis in six ancient Tuscan varieties, *Sci. Rep.* 9 (1) (2019) 3557.
- [20] A. Zerva, C. Pentari, A. Termentzi, A.H.P. America, D. Zouraris, S. K. Bhattacharya, A. Karantonis, G.I. Zervakis, E. Topakas, Discovery of two novel laccase-like multicopper oxidases from *Pleurotus citrinopileatus* and their application in phenolic oligomer synthesis, *Biotechnol. Biofuels* 14 (1) (2021) 83.
- [21] M. Valles, A.F. Kamaruddin, L.S. Wong, C.F. Blanford, Inhibition in multicopper oxidases: a critical review, *Catal. Sci. Technol.* 10 (16) (2020) 5386–5410.
- [22] A.O. Falade, U.U. Nwodo, B.C. Iweriebior, E. Green, L.V. Mabinya, A.I. Okoh, Lignin peroxidase functionalities and prospective applications, *MicrobiologyOpen* 6 (1) (2017).
- [23] X. Qin, H. Luo, X. Zhang, B. Yao, F. Ma, X. Su, Dye-decolorizing peroxidases in *Irpea lacteus* combining the catalytic properties of heme peroxidases and laccase play important roles in ligninolytic system, *Biotechnol. Biofuels* 11 (1) (2018) 302.
- [24] H. Xu, M.-Y. Guo, Y.-H. Gao, X.-H. Bai, X.-W. Zhou, Expression and characteristics of manganese peroxidase from *Ganoderma lucidum* in *Pichia pastoris* and its application in the degradation of four dyes and phenol, *BMC Biotechnol.* 17 (1) (2017) 19.
- [25] Y. Chang, D. Yang, R. Li, T. Wang, Y. Zhu, Textile dye biodecolorization by manganese peroxidase: a review, *Molecules* (2021).
- [26] K. Athamneh, A. Alneyadi, A. Alsadik, T.S. Wong, S.S. Ashraf, Efficient degradation of various emerging pollutants by wild type and evolved fungal DyP4 peroxidases, *PLoS One* 17 (1) (2022), e0262492.
- [27] K.G. Welinder, Superfamily of plant, fungal and bacterial peroxidases, *Curr. Opin. Struct. Biol.* 2 (3) (1992) 388–393.
- [28] G. de Gonzalo, D.I. Colpa, M.H.M. Habib, M.W. Fraaije, Bacterial enzymes involved in lignin degradation, *J. Biotechnol.* 236 (2016) 110–119.
- [29] M.H. Habib, H.J. Rozeboom, M.W. Fraaije, Characterization of a new DyP-peroxidase from the alkaliphilic cellulomonad, *Cellulomonas bogoriensis*, *Molecules* (Basel, Switzerland) 24 (7) (2019) 1208.
- [30] R. Singh, L.D. Eltis, The multihued palette of dye-decolorizing peroxidases, *Arch. Biochem. Biophys.* 574 (2015) 56–65.
- [31] T. Yoshida, Y. Sugano, A structural and functional perspective of DyP-type peroxidase family, *Arch. Biochem. Biophys.* 574 (2015) 49–55.
- [32] T.M. D'Souza, C.S. Merritt, C.A. Reddy, Lignin-modifying enzymes of the white rot basidiomycete *Ganoderma lucidum*, *Appl. Environ. Microbiol.* 65 (12) (1999) 5307–5313.
- [33] V. Elisashvili, E. Kachlishvili, M.D. Asatiani, R. Darlington, K.H. Kucharzyk, Physiological peculiarities of lignin-modifying enzyme production by the white-rot Basidiomycete *Coriolopsis gallica* strain BCC 142, *Microorganisms* (2017).
- [34] L. Levin, F. Forchiassin, A.M. Ramos, Copper induction of lignin-modifying enzymes in the white-rot fungus *Trametes trogii*, *Mycologia* 94 (3) (2002) 377–383.
- [35] Y.S. Yang, J.T. Zhou, H. Lu, Y.L. Yuan, L.H. Zhao, Isolation and characterization of a fungus *Aspergillus* sp. strain F-3 capable of degrading alkali lignin, *Biodegradation* 22 (5) (2011) 1017–1027.
- [36] S. Sun, S. Xie, Y. Cheng, H. Yu, H. Zhao, M. Li, X. Li, X. Zhang, J.S. Yuan, S.Y. Dai, Enhancement of environmental hazard degradation in the presence of lignin: a proteomics study, *Sci. Rep.* 7 (1) (2017) 11356.
- [37] L.D.M. Sahadevan, C.S. Misra, V. Thankamani, Characterization of lignin-degrading enzymes (LDEs) from a dimorphic novel fungus and identification of products of enzymatic breakdown of lignin, *3 Biotech* 6 (1) (2016) 56.
- [38] T.M. D'Souza, C.S. Merritt, C.A. Reddy, Lignin-modifying enzymes of the white rot basidiomycete *Ganoderma lucidum*, *Appl. Environ. Microbiol.* 65 (12) (1999) 5307–5313.
- [39] C. Qi-he, S. Krügener, T. Hirth, S. Rupp, S. Zibek, Co-cultured production of lignin-modifying enzymes with white-rot fungi, *Appl. Biochem. Biotechnol.* 165 (2) (2011) 700–718.
- [40] C. del Cerro, E. Erickson, T. Dong, R. Wong Allison, C. Eder Elizabeth, O. Purvine Samuel, D. Mitchell Hugh, K. Weitz Karl, M. Markkille Lye, C. Burnet Meagan, W. Hoyt David, K. Chu Rosalie, J.-F. Cheng, J. Ramirez Kelsey, R. Katahira, W. Xiong, E. Himmel Michael, V. Subramanian, G. Linger Jeffrey, D. Salvachúa, Intracellular pathways for lignin catabolism in white-rot fungi, *Proc. Natl. Acad. Sci.* 118 (9) (2021), e2017381118.
- [41] I. Ayuso-Fernández, J. Rencoret, A. Gutiérrez, J. Ruiz-Dueñas Francisco, T. Martínez Angel, Peroxidase evolution in white-rot fungi follows wood lignin evolution in plants, *Proc. Natl. Acad. Sci.* 116 (36) (2019) 17900–17905.
- [42] D.V. Vasina, K.V. Moiseenko, T.V. Fedorova, T.V. Tyazhelova, Lignin-degrading peroxidases in white-rot fungus *Trametes hirsuta* 072. Absolute expression quantification of full multigene family, *PLoS One* 12 (3) (2017), e0173813.
- [43] G. Songulashvili, G.A. Jimenez-Tobón, C. Jaspers, M.J. Penninckx, Immobilized laccase of *Cerrena unicolor* for elimination of endocrine disruptor micropollutants, *Fungal Biol.* 116 (8) (2012) 883–889.
- [44] M.S. Revankar, S.S. Lele, Enhanced production of laccase using a new isolate of white rot fungus WR-1, *Process Biochem.* 41 (3) (2006) 581–588.
- [45] E. Kachlishvili, E. Metreveli, V. Elisashvili, Modulation of *Cerrena unicolor* laccase and manganese peroxidase production, *SpringerPlus* 3 (1) (2014) 1–7.
- [46] C. Galhaup, H. Wagner, B. Hinterstoisser, D. Haltrich, Increased production of laccase by the wood-degrading basidiomycete *Trametes pubescens*, *Enzym. Microb. Technol.* 30 (4) (2002) 529–536.
- [47] A.N. Kapich, B.A. Prior, A. Botha, S. Galkin, T. Lundell, A. Hatakka, Effect of lignocellulose-containing substrates on production of ligninolytic peroxidases in submerged cultures of *Phanerochaete chrysosporium* ME-446, *Enzym. Microb. Technol.* 34 (2) (2004) 187–195.
- [48] A.B. Orth, D.J. Royse, M. Tien, Ubiquity of lignin-degrading peroxidases among various wood-degrading fungi, *Appl. Environ. Microbiol.* 59 (12) (1993) 4017–4023.
- [49] P.D. Patil, G.D. Yadav, Comparative studies of white-rot fungal strains (*Trametes hirsuta* MTCC-1171 and *Phanerochaete chrysosporium* NCIM-1106) for effective degradation and bioconversion of ferulic acid, *ACS Omega* 3 (11) (2018) 14858–14868.
- [50] J. Singh, A. Das, K.N. Yogalakshmi, Enhanced laccase expression and azo dye decolorization during co-interaction of *Trametes versicolor* and *Phanerochaete chrysosporium*, *SN Appl. Sci.* 2 (6) (2020) 1095.
- [51] N. Lončar, N. Drašković, N. Božić, E. Romero, S. Simić, Z. Vujčić, M. W. Fraaije, Expression and characterization of a dye-decolorizing peroxidase from *Pseudomonas fluorescens* Pf0-1, *Catalysts* 9 (5) (2019).

- [52] A.T.N. Dao, M. Smits, H.T.C. Dang, A. Brouwer, T.E. de Boer, Elucidating fungal Rigidoporus species FMD21 lignin-modifying enzyme genes and 2,3,7,8-tetrachlorodibenzo-p-dioxin degradation by laccase isozymes, *Enzym. Microb. Technol.* 147 (2021), 109800.
- [53] A.T.N. Dao, S.J. Loenen, K. Swart, H.T.C. Dang, A. Brouwer, T.E. de Boer, Characterization of 2,3,7,8-tetrachlorodibenzo-p-dioxin biodegradation by extracellular lignin-modifying enzymes from ligninolytic fungus, *Chemosphere* 263 (2021), 128280.
- [54] A.F. Ariste, R.A. Batista-García, V.K. Vaidyanathan, N. Raman, V. K. Vaithyanathan, J.L. Folch-Mallol, S.A. Jackson, A.D.W. Dobson, H. Cabana, Mycoremediation of phenols and polycyclic aromatic hydrocarbons from a biorefinery wastewater and concomitant production of lignin modifying enzymes, *J. Clean. Prod.* 253 (2020), 119810.
- [55] A.T.N. Dao, J. Vonck, T.K.S. Janssens, H.T.C. Dang, A. Brouwer, T.E. de Boer, Screening white-rot fungi for bioremediation potential of 2,3,7,8-tetrachlorodibenzo-p-dioxin, *Ind. Crop. Prod.* 128 (2019) 153–161.
- [56] D.K. Joshi, M.H. Gold, Oxidation of dibenzo-p-dioxin by lignin peroxidase from the basidiomycete *Phanerochaete chrysosporium*, *Biochemistry* 33 (36) (1994) 10969–10976.
- [57] P.O. Bankole, A.A. Adekunle, B.-H. Jeon, S.P. Govindwar, Novel cobiomass degradation of NSAIDs by two wood rot fungi, *Ganoderma applanatum* and *Laetiporus sulphureus*: ligninolytic enzymes induction, isotherm and kinetic studies, *Ecotoxicol. Environ. Saf.* 203 (2020), 110997.
- [58] Y. Shi, L. Chai, C. Tang, Z. Yang, Y. Zheng, Y. Chen, Q. Jing, Biochemical investigation of kraft lignin degradation by *Pandoraea* sp. B-6 isolated from bamboo slips, *Bioprocess Biosyst. Eng.* 36 (12) (2013) 1957–1965.
- [59] A.K. Singh, P. Yadav, R.N. Bharagava, G.D. Saratale, A. Raj, Biotransformation and cytotoxicity evaluation of kraft lignin degraded by ligninolytic *Serratia liquefaciens*, *Front. Microbiol.* 10 (2019).
- [60] Y.H. Chen, L.Y. Chai, Y.H. Zhu, Z.H. Yang, Y. Zheng, H. Zhang, Biodegradation of kraft lignin by a bacterial strain *Comamonas* sp. B-9 isolated from eroded bamboo slips, *J. Appl. Microbiol.* 112 (5) (2012) 900–906.
- [61] W. Zhang, W. Wang, J. Wang, G. Shen, Y. Yuan, L. Yan, H. Tang, W. Wang, Isolation and characterization of a novel laccase for lignin degradation, *LacZ1*, *Appl. Environ. Microbiol.* 87 (23) (2021), e0135521.
- [62] M. Kumar, J. Singh, M.K. Singh, A. Singhal, I.S. Thakur, Investigating the degradation process of kraft lignin by  $\beta$ -proteobacterium, *Pandoraea* sp. ISTKB, *Environ. Sci. Pollut. Res. Int.* 22 (20) (2015) 15690–15702.
- [63] Y.I. Xiong, Y. Zhao, K. Ni, Y. Shi, Q. Xu, Characterization of ligninolytic bacteria and analysis of alkali-lignin biodegradation products, *Pol. J. Microbiol.* 69 (3) (2020) 339–347.
- [64] H.T. Imam, P.C. Marr, A.C. Marr, Enzyme entrapment, biocatalyst immobilization without covalent attachment, *Green Chem.* 23 (14) (2021) 4980–5005.
- [65] P. Nandal, A. Arora, S. Virmani, An appraisal on valorization of lignin: a byproduct from biorefineries and paper industries, *Biomass Bioenergy* 155 (2021), 106295.
- [66] A. Haile, G.G. Gelebo, T. Tesfaye, W. Mengie, M.A. Mebrate, A. Abuhay, D. Y. Limeneh, Pulp and paper mill wastes: utilizations and prospects for high value-added biomaterials, *Bioresour. Bioprocess.* 8 (1) (2021) 35.
- [67] T. Li, S. Takkellapati, The current and emerging sources of technical lignins and their applications, *Biofuels Bioprod. Biorefin.* 12 (5) (2018) 756–787.
- [68] R.R. Singhania, A.K. Patel, T. Raj, C.-W. Chen, V.K. Ponnusamy, N. Tahir, S.-H. Kim, C.-D. Dong, Lignin valorisation via enzymes: a sustainable approach, *Fuel* 311 (2022), 122608.
- [69] M.E. Eugenio, D. Ibarra, R. Martín-Sampedro, E. Espinosa, I. Bascón, A. Rodríguez, Alternative raw materials for pulp and paper production in the concept of a lignocellulosic biorefinery, *Cellulose* 12 (2019) 78.
- [70] C. Weng, X. Peng, Y. Han, Depolymerization and conversion of lignin to value-added bioproducts by microbial and enzymatic catalysis, *Biotechnol. Biofuels* 14 (1) (2021) 84.
- [71] J. Zakzeski, P.C.A. Bruijninx, A.L. Jongerius, B.M. Weckhuysen, The catalytic valorization of lignin for the production of renewable chemicals, *Chem. Rev.* 110 (6) (2010) 3552–3599.
- [72] J. Dillies, C. Vivien, M. Chevalier, A. Rulence, G. Châtaigné, C. Flahaut, V. Senez, R. Froidevaux, Enzymatic depolymerization of industrial lignins by laccase-mediator systems in 1,4-dioxane/water, *Biotechnol. Appl. Biochem.* 67 (5) (2020) 774–782.
- [73] A.K. Singh, M. Bilal, H.M.N. Iqbal, A. Raj, Trends in predictive biodegradation for sustainable mitigation of environmental pollutants: recent progress and future outlook, *Sci. Total Environ.* 770 (2021), 144561.
- [74] A.K. Singh, M. Bilal, H.M.N. Iqbal, A. Raj, *In silico* analytical toolset for predictive degradation and toxicity of hazardous pollutants in water sources, *Chemosphere* 292 (2022), 133250.
- [75] A.K. Singh, S.K. Katari, A. Umamaheswari, A. Raj, *In silico* exploration of lignin peroxidase for unraveling the degradation mechanism employing lignin model compounds, *RSC Adv.* 11 (24) (2021) 14632–14653.
- [76] O. Benslama, N. Mansouri, R. Arhab, *In silico* investigation of the lignin polymer biodegradation by two actinomycetal peroxidase enzymes, *Mater. Today Proc.* 53 (2022) 1–5.
- [77] A.K.S. Kameshwar, R. Barber, W. Qin, Comparative modeling and molecular docking analysis of white, brown and soft rot fungal laccases using lignin model compounds for understanding the structural and functional properties of laccases, *J. Mol. Graph. Model.* 79 (2018) 15–26.
- [78] P. Bhatt, K. Bhatt, W.-J. Chen, Y. Huang, Y. Xiao, S. Wu, Q. Lei, J. Zhong, X. Zhu, S. Chen, Bioremediation potential of laccase for catalysis of glyphosate, isoproturon, lignin, and parathion: molecular docking, dynamics, and simulation, *J. Hazard. Mater.* 443 (2023), 130319.
- [79] A.K. Singh, M. Bilal, T. Jesionowski, H.M.N. Iqbal, Deployment of oxidoreductases for sustainable biocatalytic degradation of selected endocrine-disrupting chemicals, *Sustain. Chem. Pharm.* 31 (2023), 100934.
- [80] A.K. Singh, M. Bilal, T. Jesionowski, H.M.N. Iqbal, Assessing chemical hazard and unraveling binding affinity of priority pollutants to lignin modifying enzymes for environmental remediation, *Chemosphere* 313 (2023), 137546.
- [81] G. Janusz, A. Pawlik, J. Sulej, U. Swiderska-Burek, A. Jarosz-Wilkolazka, A. Paszczynski, Lignin degradation: microorganisms, enzymes involved, genomes analysis and evolution, *FEMS Microbiol. Rev.* 41 (6) (2017) 941–962.
- [82] R. Rahmanpour, T.D.H. Bugg, Characterisation of Dyp-type peroxidases from *Pseudomonas fluorescens* Pf-5: oxidation of Mn(II) and polymeric lignin by Dyp1B, *Arch. Biochem. Biophys.* 574 (2015) 93–98.
- [83] T.D.H. Bugg, R. Rahmanpour, Enzymatic conversion of lignin into renewable chemicals, *Curr. Opin. Chem. Biol.* 29 (2015) 10–17.
- [84] D. Grgas, M. Rukavina, D. Beško, T. Štefanac, V. Crnek, T. Šikić, M. Habuda-Stanić, T. Landeka Dragičević, The bacterial degradation of lignin—a review, *Water* (2023).
- [85] J.-C. Sigoillot, J.-G. Berrin, M. Bey, L. Lesage-Meessen, A. Levasseur, A. Lomascolo, E. Record, E. Uzan-Boukhris, Chapter 8 - fungal strategies for lignin degradation, in: L. Jouanin, C. Lapiere (Eds.), *Advances in Botanical Research*, Academic Press, 2012, pp. 263–308.
- [86] T.D. Bugg, M. Ahmad, E.M. Hardiman, R. Singh, The emerging role for bacteria in lignin degradation and bio-product formation, *Curr. Opin. Biotechnol.* 22 (3) (2011) 394–400.
- [87] N. Kamimura, K. Takahashi, K. Mori, T. Araki, M. Fujita, Y. Higuchi, E. Masai, Bacterial catabolism of lignin-derived aromatics: new findings in a recent decade: update on bacterial lignin catabolism, *Environ. Microbiol. Rep.* 9 (6) (2017) 679–705.
- [88] C. del Cerro, E. Erickson, T. Dong, A.R. Wong, E.K. Eder, S.O. Purvine, H. D. Mitchell, K.K. Weitz, L.M. Markillie, M.C. Burnet, D.W. Hoyt, R.K. Chu, J.-F. Cheng, K.J. Ramirez, R. Katahira, W. Xiong, M.E. Himmel, V. Subramanian, J. G. Linger, D. Salvachúa, Intracellular pathways for lignin catabolism in white-rot fungi, *Proc. Natl. Acad. Sci.* 118 (9) (2021), e2017381118.
- [89] F. Martani, F. Beltrametti, D. Porro, P. Branduardi, M. Lotti, The importance of fermentation conditions for the biotechnological production of lignin modifying enzymes from white-rot fungi, *FEMS Microbiol. Lett.* 364 (13) (2017) fnx134.
- [90] J.E. Kowalczyk, M. Peng, M. Pawlowski, A. Lipzen, V. Ng, V. Singan, M. Wang, I. V. Grigoriev, M.R. Mäkelä, The white-rot basidiomycete *Dichomitus squalens* shows highly specific transcriptional response to lignocellulose-related aromatic compounds, *Front. Bioeng. Biotechnol.* 7 (2019).
- [91] K. Peraza-Jiménez, S. De la Rosa-García, J.J. Hujjara-Vasconcelos, M. Reyes-Estebanez, S. Gómez-Cornelio, Enzymatic bioprospecting of fungi isolated from a tropical rainforest in Mexico, *J. Fungi* (2022).
- [92] A.T. Martínez, M. Speranza, F.J. Ruiz-Dueñas, P. Ferreira, S. Camarero, F. Guillén, M.J. Martínez, A. Gutiérrez, J.C. del Río, Biodegradation of lignocelluloses: microbial, chemical, and enzymatic aspects of the fungal attack of lignin, *Int. Microbiol.* 8 (3) (2005) 195–204.
- [93] E. Veloz Villavicencio, T. Mali, H.K. Mattila, T. Lundell, Enzyme activity profiles produced on wood and straw by four fungi of different decay strategies, *Microorganisms* 8 (1) (2020).
- [94] W. Zhang, X. Ren, Q. Lei, L. Wang, Screening and comparison of lignin degradation microbial consortia from wooden antiques, *Molecules* 26 (10) (2021).
- [95] B.R. Kang, S.Y. Kim, M. Kang, T.K. Lee, Removal of pharmaceuticals and personal care products using native fungal enzymes extracted during the ligninolytic process, *Environ. Res.* 195 (2021), 110878.
- [96] S. Bao, Z. Teng, S. Ding, Heterologous expression and characterization of a novel laccase isoenzyme with dyes decolorization potential from *Coprinus comatus*, *Mol. Biol. Rep.* 40 (2) (2013) 1927–1936.
- [97] D. Daassi, S. Rodríguez-Couto, M. Nasri, T. Mechichi, Biodegradation of textile dyes by immobilized laccase from *Coriopsis gallica* into Ca-alginate beads, *Int. Biodeterior. Biodegradation* 90 (2014) 71–78.
- [98] C. Junghanns, G. Krauss, D. Schlosser, Potential of aquatic fungi derived from diverse freshwater environments to decolorise synthetic azo and anthraquinone dyes, *Bioresour. Technol.* 99 (5) (2008) 1225–1235.
- [99] M. Chhabra, S. Mishra, T.R. Sreekrishnan, Mediator-assisted decolorization and detoxification of textile dyes/dye mixture by *Cyathus bulleri* laccase, *Appl. Biochem. Biotechnol.* 151 (2) (2008) 587–598.
- [100] R.T. Mehmood, M.J. Asad, M. Asgher, S.H. Hadri, M. Gulfranz, J.D. Wu, M. I. Bhatti, N. Zaman, D. Ahmed, First report of using response surface methodology for the biodegradation of single azo disperse dyes by indigenous *Daedalea dickinsii*-IEBL-2, *Rom. Biotechnol. Lett.* 25 (2020) 1236–1245.
- [101] M.C. Abrahão, A.d.M. Gugliotta, R. Da Silva, R.J.Y. Fujieda, M. Boscolo, E. Gomes, Ligninolytic activity from newly isolated basidiomycete strains and effect of these enzymes on the azo dye orange II decolorisation, *Ann. Microbiol.* 58 (3) (2008) 427–432.
- [102] L.F. Longe, J. Couvreur, M. Leriche Grandchamp, G. Garnier, F. Allais, K. Saito, Importance of mediators for lignin degradation by fungal laccase, *ACS Sustain. Chem. Eng.* 6 (8) (2018) 10097–10107.
- [103] X. Qin, J. Zhang, X. Zhang, Y. Yang, Induction, purification and characterization of a novel manganese peroxidase from *Irpex lacteus* CD2 and its application in the decolorization of different types of dye, *PLoS One* 9 (11) (2014), e113282.

- [104] V. Kokol, A. Doliška, I. Eichlerová, P. Baldrian, F. Nerud, Decolorization of textile dyes by whole cultures of *Ischnoderma resinosa* and by purified laccase and Mn-peroxidase, *Enzym. Microb. Technol.* 40 (7) (2007) 1673–1677.
- [105] R.K. Pandey, S. Tewari, L. Tewari, Lignolytic mushroom *Lenzites elegans* WDP2: laccase production, characterization, and bioremediation of synthetic dyes, *Ecotoxicol. Environ. Saf.* 158 (2018) 50–58.
- [106] D. Iark, A.J. dos Reis Buzzo, J.A.A. Garcia, V.G. Correa, C.V. Helm, R.C.G. Correa, R.A. Peralta, R.d.F.P.M. Moreira, A. Bracht, R.M. Peralta, Enzymatic degradation and detoxification of azo dye Congo red by a new laccase from *Oudemansiella canarii*, *Bioresour. Technol.* 289 (2019), 121655.
- [107] L.B. Brenelli, G.F. Persinoti, J.P.L.F. Cairo, M.V. Liberato, T.A. Gonçalves, I.V. R. Otero, P.H. Mainardi, C. Felby, L.D. Sette, F.M. Squina, Novel redox-active enzymes for ligninolytic applications revealed from multiomics analyses of *Peniophora* sp. CBMAI 1063, a laccase hyper-producer strain, *Sci. Rep.* 9 (1) (2019) 17564.
- [108] E. Krumova, N. Kostadinova, J. Miteva-Staleva, G. Stoyancheva, B. Spassova, R. Abrashev, M. Angelova, Potential of ligninolytic enzymatic complex produced by white-rot fungi from genus *Trametes* isolated from Bulgarian forest soil, *Eng. Life Sci.* 18 (9) (2018) 692–701.
- [109] P. Permpornsakul, S. Prasongsuk, P. Lotrakul, D.E. Eveleigh, D.Y. Kobayashi, T. Imai, H. Punnapayak, Biological treatment of reactive Black 5 by resupinate white rot fungus *Phanerochaete sordida* PBU 0057, *Pol. J. Environ. Res.* 25 (3) (2016).
- [110] R. Cruz-Ornelas, J.E. Sánchez-Vázquez, L. Amaya-Delgado, K. Guillén-Navarro, A. Calixto-Romo, Biodegradation of NSAIDs and their effect on the activity of ligninolytic enzymes from *Pleurotus djamor*, *3 Biotech* 9 (10) (2019) 373.
- [111] Y. Gao, J.-J. Li, L. Zheng, Y. Du, Rational design of *Pleurotus eryngii* versatile ligninolytic peroxidase for enhanced pH and thermal stability through structure-based protein engineering, *Protein Eng. Des. Sel.* 30 (11) (2017) 743–751.
- [112] D. Knop, D. Levinson, A. Makovitzki, A. Agami, E. Lerer, A. Mimran, O. Yarden, Y. Hadar, A.A. Brakhage, Limits of versatility of versatile peroxidase, *Appl. Environ. Microbiol.* 82 (14) (2016) 4070–4080.
- [113] D. Knop, J. Ben-Ari, T.M. Salame, D. Levinson, O. Yarden, Y. Hadar, Mn<sup>2+</sup>-deficiency reveals a key role for the *Pleurotus ostreatus* versatile peroxidase (VP4) in oxidation of aromatic compounds, *Appl. Microbiol. Biotechnol.* 98 (15) (2014) 6795–6804.
- [114] I. Bibi, S. Javed, S. Ata, F. Majid, S. Kamal, M. Sultan, K. Jilani, M. Umair, M. I. Khan, M. Iqbal, Biodegradation of synthetic orange G dye by *Pleurotus sajor caju* with *Punica granatum* peel as natural mediator, *Biocatal. Agric. Biotechnol.* 22 (2019), 101420.
- [115] R.S. Yehia, S. Rodriguez-Couto, Discoloration of the azo dye Congo Red by manganese-dependent peroxidase from *Pleurotus sajor caju*, *Appl. Biochem. Microbiol.* 53 (2) (2017) 222–229.
- [116] J. Juárez-Hernández, D. Castillo-Hernández, C. Pérez-Parada, S. Nava-Galicia, J. A. Cuervo-Parra, E. Surian-Cruz, G. Díaz-Godínez, C. Sánchez, M. Bibbins-Martínez, Isolation of fungi from a textile industry effluent and the screening of their potential to degrade industrial dyes, *J. Fungi* (2021).
- [117] V. Hahn, M. Meister, S. Hussy, A. Cordes, G. Enderle, A. Saningong, F. Schauer, Enhanced laccase-mediated transformation of diclofenac and flufenamic acid in the presence of bisphenol A and testing of an enzymatic membrane reactor, *AMB Express* 8 (1) (2018) 28.
- [118] G.M.M. Rashid, T.D.H. Bugg, Enhanced biocatalytic degradation of lignin using combinations of lignin-degrading enzymes and accessory enzymes, *Catal. Sci. Technol.* 11 (10) (2021) 3568–3577.
- [119] Z. Xu, L. Qin, M. Cai, W. Hua, M. Jin, Biodegradation of kraft lignin by newly isolated *Klebsiella pneumoniae*, *Pseudomonas putida*, and *Ochrobactrum tritici* strains, *Environ. Sci. Pollut. Res.* 25 (14) (2018) 14171–14181.
- [120] A. Blánquez, A.S. Ball, J.A. González-Pérez, N.T. Jiménez-Morillo, F. González-Vila, M.E. Arias, M. Hernández, Laccase SilA from *Streptomyces ipomoeae* CECT 3341, a key enzyme for the degradation of lignin from agricultural residues? *PLoS One* 12 (11) (2017), e0187649.
- [121] C. Chen, R. Shrestha, K. Jia, P.F. Gao, B.V. Geisbrecht, S.H. Bossmann, J. Shi, P. Li, Characterization of dye-decolorizing peroxidase (DyP) from *Thermomonospora curvata* reveals unique catalytic properties of A-type DyPs, *J. Biol. Chem.* 290 (38) (2015) 23447–23463.
- [122] C. Barbosa, C.M. Silveira, D. Silva, V. Brissos, P. Hildebrandt, L.O. Martins, S. Todorovic, Immobilized dye-decolorizing peroxidase (DyP) and directed evolution variants for hydrogen peroxide biosensing, *Biosens. Bioelectron.* 153 (2020), 112055.
- [123] S. Kim, Mushroom ligninolytic enzymes—features and application of potential enzymes for conversion of lignin into bio-based chemicals and materials, *Appl. Sci.* 11 (13) (2021).
- [124] The Protein Data Bank (PDB). <https://www.rcsb.org>. (Accessed 21 May 2023).
- [125] H.M. Berman, J. Westbrook, Z. Feng, G. Gilliland, T.N. Bhat, H. Weissig, I. N. Shindyalov, P.E. Bourne, The Protein Data Bank, *Nucleic Acids Res.* 28 (1) (2000) 235–242.
- [126] Z. Duan, R. Shen, B. Liu, M. Yao, R. Jia, Comprehensive investigation of a dye-decolorizing peroxidase and a manganese peroxidase from *Irpelex lacteus* F17, a lignin-degrading basidiomycete, *AMB Express* 8 (1) (2018) 119.
- [127] F. Martani, F. Beltrametti, D. Porro, P. Branduardi, M. Lotti, The importance of fermentative conditions for the biotechnological production of lignin modifying enzymes from white-rot fungi, *FEMS Microbiol. Lett.* 364 (13) (2017).
- [128] C.J. Houtman, E. Maligaspe, C.G. Hunt, E. Fernandez-Fueyo, A.T. Martínez, K. E. Hammel, Fungal lignin peroxidase does not produce the veratryl alcohol cation radical as a diffusible ligninolytic oxidant, *J. Biol. Chem.* 293 (13) (2018) 4702–4712.
- [129] X. Qin, X. Sun, H. Huang, Y. Bai, Y. Wang, H. Luo, B. Yao, X. Zhang, X. Su, Oxidation of a non-phenolic lignin model compound by two *Irpelex lacteus* manganese peroxidases: evidence for implication of carboxylate and radicals, *Biotechnol. Biofuels* 10 (1) (2017) 103.
- [130] L.T.M. Pham, K. Deng, T.R. Northen, S.W. Singer, P.D. Adams, B.A. Simmons, K. L. Sale, Experimental and theoretical insights into the effects of pH on catalysis of bond-cleavage by the lignin peroxidase isozyme H8 from *Phanerochaete chrysosporium*, *Biotechnol. Biofuels* 14 (1) (2021) 108.
- [131] H. Son, H. Seo, S. Han, S.M. Kim, L.T.M. Pham, M.F. Khan, H.J. Sung, S.-h. Kang, K.-J. Kim, Y.H. Kim, Extra disulfide and ionic salt bridge improves the thermostability of lignin peroxidase H8 under acidic condition, *Enzym. Microb. Technol.* 148 (2021), 109803.
- [132] W. Blodig, A.T. Smith, W.A. Doyle, K. Piontek, Crystal structures of pristine and oxidatively processed lignin peroxidase expressed in *Escherichia coli* and of the W171F variant that eliminates the redox active tryptophan 171. Implications for the reaction mechanism1 Edited by R. Huber, *J. Mol. Biol.* 305 (4) (2001) 851–861.
- [133] T. Choinowski, W. Blodig, K.H. Winterhalter, K. Piontek, The crystal structure of lignin peroxidase at 1.70 Å resolution reveals a hydroxy group on the C $\beta$  of tryptophan 171: a novel radical site formed during the redox cycle1 Edited by R. Huber, *J. Mol. Biol.* 286 (3) (1999) 809–827.
- [134] H.M. Berman, J. Westbrook, Z. Feng, G. Gilliland, T.N. Bhat, H. Weissig, I. N. Shindyalov, P.E. Bourne, The Protein Data Bank, *Nucleic Acids Res.* 28 (1) (2000) 235–242.
- [135] Y. Miki, F.R. Calviño, R. Pogni, S. Giansanti, F.J. Ruiz-Dueñas, M.J. Martínez, R. Basosi, A. Romero, A.T. Martínez, Crystallographic, kinetic, and spectroscopic study of the first ligninolytic peroxidase presenting a catalytic tyrosine, *J. Biol. Chem.* 286 (17) (2011) 15525–15534.
- [136] C. Geourjon, G. Deléage, SOPMA: significant improvements in protein secondary structure prediction by consensus prediction from multiple alignments, *Bioinformatics* 11 (6) (1995) 681–684.
- [137] E. Gasteiger, A. Gattiker, C. Hoogland, I. Ivanyi, R.D. Appel, A. Bairoch, ExPASy: the proteomics server for in-depth protein knowledge and analysis, *Nucleic Acids Res.* 31 (13) (2003) 3784–3788.
- [138] J.H. Pereira, R.A. Heins, D.L. Gall, R.P. McAndrew, K. Deng, K.C. Holland, T. J. Donohue, D.R. Noguera, B.A. Simmons, K.L. Sale, J. Ralph, P.D. Adams, Structural and biochemical characterization of the early and late enzymes in the lignin  $\beta$ -aryl ether cleavage pathway from *Sphingobium* sp. SYK-6, *J. Biol. Chem.* 291 (19) (2016) 10228–10238.
- [139] D.W.S. Wong, Structure and action mechanism of ligninolytic enzymes, *Appl. Biochem. Biotechnol.* 157 (2) (2009) 174–209.
- [140] M. Tien, T.K. Kirk, Lignin peroxidase of *Phanerochaete chrysosporium*, in: *Methods in Enzymology*, Elsevier, 1988, pp. 238–249.
- [141] F.A. Riyadi, A.A. Tahir, N. Yusof, N.S.A. Sabri, M.J.M.M. Noor, F.N.M.D. Akhir, N. A. Othman, Z. Zakaria, H. Hara, Enzymatic and genetic characterization of lignin depolymerization by *Streptomyces* sp. S6 isolated from a tropical environment, *Sci. Rep.* 10 (1) (2020) 7813.
- [142] G.K. Parshetti, S. Parshetti, D.C. Kalyani, R.-a. Doong, S.P. Govindwar, Industrial dye decolorizing lignin peroxidase from *Kocuria rosea* MTCC 1532, *Ann. Microbiol.* 62 (1) (2012) 217–223.
- [143] A.O. Falade, U.U. Nwodo, B.C. Iweriebor, E. Green, L.V. Mabinya, A.I. Okoh, Lignin peroxidase functionalities and prospective applications, *Microbiol. Biotechnol.* 6 (1) (2017), e00394.
- [144] R.T. Lamar, The role of fungal lignin-degrading enzymes in xenobiotic degradation, *Curr. Opin. Biotechnol.* 3 (3) (1992) 261–266.
- [145] H. Hirai, M. Sugiura, S. Kawai, T. Nishida, Characteristics of novel lignin peroxidases produced by white-rot fungus *Phanerochaete sordida* YK-624, *FEMS Microbiol. Lett.* 246 (1) (2005) 19–24.
- [146] C.J. Houtman, E. Maligaspe, C.G. Hunt, E. Fernández-Fueyo, A.T. Martínez, K. E. Hammel, Fungal lignin peroxidase does not produce the veratryl alcohol cation radical as a diffusible ligninolytic oxidant, *J. Biol. Chem.* 293 (13) (2018) 4702–4712.
- [147] V. Brissos, D. Tavares, A.C. Sousa, M.P. Robalo, L.O. Martins, Engineering a bacterial DyP-type peroxidase for enhanced oxidation of lignin-related phenolics at alkaline pH, *ACS Catal.* 7 (5) (2017) 3454–3465.
- [148] O.D.V. Biko, M. Viljoen-Bloom, W.H. van Zyl, Microbial lignin peroxidases: applications, production challenges and future perspectives, *Enzym. Microb. Technol.* 141 (2020), 109669.
- [149] E. Fernández-Fueyo, F.J. Ruiz-Dueñas, A.T. Martínez, Engineering a fungal peroxidase that degrades lignin at very acidic pH, *Biotechnol. Biofuels* 7 (1) (2014) 114.
- [150] L.T.M. Pham, S.J. Kim, Y.H. Kim, Improvement of catalytic performance of lignin peroxidase for the enhanced degradation of lignocellulose biomass based on the imbedded electron-relay in long-range electron transfer route, *Biotechnol. Biofuels* 9 (1) (2016) 247.
- [151] A.I. Cañas, S. Camarero, Laccases and their natural mediators: biotechnological tools for sustainable eco-friendly processes, *Biotechnol. Adv.* 28 (6) (2010) 694–705.
- [152] L. Munk, M.L. Andersen, A.S. Meyer, Direct rate assessment of laccase catalysed radical formation in lignin by electron paramagnetic resonance spectroscopy, *Enzym. Microb. Technol.* 106 (2017) 88–96.
- [153] R. Pogni, M.C. Baratto, A. Sinicropi, R. Basosi, Spectroscopic and computational characterization of laccases and their substrate radical intermediates, *Cell. Mol. Life Sci.* 72 (5) (2015) 885–896.
- [154] R. Mehra, J. Muschiol, A.S. Meyer, K.P. Kepp, A structural-chemical explanation of fungal laccase activity, *Sci. Rep.* 8 (1) (2018) 17285.

- [155] S.M. Jones, E.I. Solomon, Electron transfer and reaction mechanism of laccases, *Cell. Mol. Life Sci.* 72 (5) (2015) 869–883.
- [156] J. Ihssen, M. Schubert, L. Thöny-Meyer, M. Richter, Laccase catalyzed synthesis of iodinated phenolic compounds with antifungal activity, *PLoS One* 9 (3) (2014), e89924.
- [157] R. Hilgers, J.-P. Vincken, H. Gruppen, M.A. Kabel, Laccase/mediator systems: their reactivity toward phenolic lignin structures, *ACS Sustain. Chem. Eng.* 6 (2) (2018) 2037–2046.
- [158] J.M. Khaled, S.A. Alyahya, R. Govindan, C.K. Chelliah, M. Maruthupandy, N. S. Alharbi, S. Kadaikunnan, R. Issac, S. Murugan, W.-J. Li, Laccase producing bacteria influenced the high decolorization of textile azo dyes with advanced study, *Environ. Res.* 207 (2022), 112211.
- [159] N. Agarwal, V.S. Solanki, A. Gacem, M.A. Hasan, B. Pare, A. Srivastava, A. Singh, V.K. Yadav, K.K. Yadav, C. Lee, W. Lee, S. Chaiyaprat, B.-H. Jeon, Bacterial laccases as biocatalysts for the remediation of environmental toxic pollutants: a green and eco-friendly approach—a review, *Water* (2022).
- [160] C.E. Edoamodu, U.U. Nwodo, Decolorization of synthetic dyes by laccase produced from *Bacillus* sp. NU2, *Biotechnol. Equip.* 36 (1) (2022) 95–106.
- [161] L. Arregui, M. Ayala, X. Gómez-Gil, G. Gutiérrez-Soto, C.E. Hernández-Luna, M. Herrera de los Santos, L. Levin, A. Rojo-Domínguez, D. Romero-Martínez, M.C. N. Sapatarrat, M.A. Trujillo-Roldán, N.A. Valdez-Cruz, Laccases: structure, function, and potential application in water bioremediation, *Microb. Cell Factories* 18 (1) (2019) 200.
- [162] M.H. Du, Z.W. Yan, Y.J. Hao, Z.T. Yan, F.L. Si, B. Chen, L. Qiao, Suppression of Laccase 2 severely impairs cuticle tanning and pathogen resistance during the pupal metamorphosis of *Anopheles sinensis* (Diptera: Culicidae), *Parasit. Vectors* 10 (1) (2017) 171.
- [163] G. Janusz, A. Pawlik, U. Świdarska-Burek, J. Polak, J. Sulej, A. Jarosz-Wilkolazka, A. Paszczyński, Laccase properties, physiological functions, and evolution, *Int. J. Mol. Sci.* 21 (3) (2020).
- [164] E. Veloz Villavicencio, T. Mali, H.K. Mattila, T. Lundell, Enzyme activity profiles produced on wood and straw by four fungi of different decay strategies, *Microorganisms* 8 (1) (2020) 73.
- [165] NCBI. [https://www.ncbi.nlm.nih.gov/protein/NP\\_651441.1?report=fasta](https://www.ncbi.nlm.nih.gov/protein/NP_651441.1?report=fasta). (Accessed 21 May 2023).
- [166] T. Xie, Z. Liu, G. Wang, Structural basis for monolignol oxidation by a maize laccase, *Nat. Plants* 6 (3) (2020) 231–237.
- [167] T. Skálová, J. Dohnálek, L.H. Østergaard, P.R. Østergaard, P. Kolenko, J. Dušková, A. Štěpánková, J. Hašek, The structure of the small laccase from *Streptomyces coelicolor* reveals a link between laccases and nitrite reductases, *J. Mol. Biol.* 385 (4) (2009) 1165–1178.
- [168] T. Bertrand, C. Jolival, P. Briozzo, E. Caminade, N. Joly, C. Madzak, C. Mougin, Crystal structure of a four-copper laccase complexed with an arylamine: insights into substrate recognition and correlation with kinetics, *Biochemistry* 41 (23) (2002) 7325–7333.
- [169] PDB ID:3CG8. <https://www.rcsb.org/structure/3CG8>.
- [170] T. Skálová, J. Dohnálek, L.H. Østergaard, P.R. Østergaard, P. Kolenko, J. Dušková, A. Štěpánková, J. Hašek, The structure of the small laccase from *Streptomyces coelicolor* reveals a link between laccases and nitrite reductases, *J. Mol. Biol.* 385 (4) (2009) 1165–1178.
- [171] T. Bertrand, C. Jolival, P. Briozzo, E. Caminade, N. Joly, C. Madzak, C. Mougin, Crystal structure of a four-copper laccase complexed with an arylamine: insights into substrate recognition and correlation with kinetics, *Biochemistry* 41 (23) (2002) 7325–7333.
- [172] T. Xie, Z. Liu, G. Wang, Structural basis for monolignol oxidation by a maize laccase, *Nat. Plants* 6 (3) (2020) 231–237.
- [173] I. Matelj, M. Alcalde, Engineering a highly thermostable high-redox potential laccase, *ACS Sustain. Chem. Eng.* 9 (29) (2021) 9632–9637.
- [174] L. Munk, A.K. Sitarz, D.C. Kalyani, J.D. Mikkelsen, A.S. Meyer, Can laccases catalyze bond cleavage in lignin? *Biotechnol. Adv.* 33 (1) (2015) 13–24.
- [175] J. Wang, J. Feng, W. Jia, S. Chang, S. Li, Y. Li, Lignin engineering through laccase modification: a promising field for energy plant improvement, *Biotechnol. Biofuels* 8 (2015) 145.
- [176] G. Janusz, A. Pawlik, U. Świdarska-Burek, J. Polak, J. Sulej, A. Jarosz-Wilkolazka, A. Paszczyński, Laccase properties, physiological functions, and evolution, *Int. J. Mol. Sci.* 21 (3) (2020).
- [177] A. Kumar, S. Ahlawat, H. Mohan, K.K. Sharma, Stabilization–destabilization and redox properties of laccases from medicinal mushroom *Ganoderma lucidum* and human pathogen *Yersinia enterocolitica*, *Int. J. Biol. Macromol.* 167 (2021) 369–381.
- [178] K. Mayolo-Delosa, M. González-González, M. Rito-Palomares, Laccases in food industry: bioprocessing, potential industrial and biotechnological applications, *Front. Bioeng. Biotechnol.* 8 (2020).
- [179] A. Zerva, S. Simić, E. Topakas, J. Nikodinovic-Runic, Applications of microbial laccases: patent review of the past decade (2009–2019), *Catalysts* (2019).
- [180] D.M. Mate, M. Alcalde, Laccase: a multi-purpose biocatalyst at the forefront of biotechnology, *Microb. Biotechnol.* 10 (6) (2017) 1457–1467.
- [181] A.I. Cañas, S. Camarero, Laccases and their natural mediators: biotechnological tools for sustainable eco-friendly processes, *Biotechnol. Adv.* 28 (6) (2010) 694–705.
- [182] T. Brugnari, D.M. Braga, C.S.A. dos Santos, B.H.C. Torres, T.A. Modkovski, C.W. I. Haminiuk, G.M. Maciel, Laccases as green and versatile biocatalysts: from lab to enzyme market—an overview, *Bioresour. Bioprocess.* 8 (1) (2021) 131.
- [183] Z.-B. Guan, Q. Luo, H.-R. Wang, Y. Chen, X.-R. Liao, Bacterial laccases: promising biological green tools for industrial applications, *Cell. Mol. Life Sci.* 75 (19) (2018) 3569–3592.
- [184] T.C. Ezike, J.O. Udeh, P.E. Joshua, A.L. Ezugwu, C.V. Isiwu, S.O.O. Eze, F. C. Chilaka, Substrate specificity of a new laccase from *Trametes polyzona* WRF03, *Heliyon* 7 (1) (2021), e06080.
- [185] Y. Chang, D. Yang, R. Li, T. Wang, Y. Zhu, Textile dye biodecolorization by manganese peroxidase: a review, *Molecules* (Basel, Switzerland) 26 (15) (2021) 4403.
- [186] K. Shi, Y. Liu, P. Chen, Y. Li, Contribution of lignin peroxidase, manganese peroxidase, and laccase in lignite degradation by mixed white-rot fungi, *Waste Biomass Valorization* (2020).
- [187] H. Rekiq, N. Zaráf Jaouadi, K. Bouacem, B. Zenati, S. Kourdali, A. Badis, R. Annane, A. Bouanane-Darenfed, S. Bejar, B. Jaouadi, Physical and enzymatic properties of a new manganese peroxidase from the white-rot fungus *Trametes pubescens* strain i8 for lignin biodegradation and textile-dyes biodecolorization, *Int. J. Biol. Macromol.* 125 (2019) 514–525.
- [188] W. Kong, H. Chen, S. Lyu, F. Ma, H. Yu, X. Zhang, Characterization of a novel manganese peroxidase from white-rot fungus *Echinodontium taxodii* 2538, and its use for the degradation of lignin-related compounds, *Process Biochem.* 51 (11) (2016) 1776–1783.
- [189] P. Lueangjaroenkit, C. Teerapatsakul, K. Sakka, M. Sakka, T. Kimura, E. Kunitake, L. Chitradon, Two manganese peroxidases and a laccase of *Trametes polyzona* KU-RNW027 with novel properties for dye and pharmaceutical product degradation in redox mediator-free system, *Mycobiology* 47 (2) (2019) 217–229.
- [190] PDB ID:3M5Q. <https://www.rcsb.org/structure/3M5Q>.
- [191] M. Sundaramoorthy, M.H. Gold, T.L. Poulos, Ultrahigh (0.93 Å) resolution structure of manganese peroxidase from *Phanerochaete chrysosporium*: implications for the catalytic mechanism, *J. Inorg. Biochem.* 104 (6) (2010) 683–690.
- [192] A. Hatakka, T. Lundell, M. Hofrichter, P. Majjala, Manganese peroxidase and its role in the degradation of wood lignin, in: *Applications of Enzymes to Lignocellulosics*, American Chemical Society, 2003, pp. 230–243.
- [193] E. Fernández-Fueyo, S. Acebes, F.J. Ruiz-Dueñas, M.J. Martínez, A. Romero, F. J. Medrano, V. Guallar, A.T. Martínez, Structural implications of the C-terminal tail in the catalytic and stability properties of manganese peroxidases from ligninolytic fungi, *Acta Crystallogr. D Biol. Crystallogr.* 70 (Pt 12) (2014) 3253–3265.
- [194] R. Shrestha, G. Huang, D.A. Meekins, B.V. Geisbrecht, P. Li, Mechanistic insights into dye-decolorizing peroxidase revealed by solvent isotope and viscosity effects, *ACS Catal.* 7 (9) (2017) 6352–6364.
- [195] G. Catucci, F. Valetti, S.J. Sadeghi, G. Gilardi, Biochemical features of dye-decolorizing peroxidases: current impact on lignin degradation, *Biotechnol. Appl. Biochem.* 67 (5) (2020) 751–759.
- [196] P. Dhankhar, V. Dalal, J.K. Mahto, B.R. Gurjar, S. Tomar, A.K. Sharma, P. Kumar, Characterization of dye-decolorizing peroxidase from *Bacillus subtilis*, *Arch. Biochem. Biophys.* 693 (2020), 108590.
- [197] D.I. Colpa, M.W. Fraaije, E. van Blois, DyP-type peroxidases: a promising and versatile class of enzymes, *J. Ind. Microbiol. Biotechnol.* 41 (1) (2014) 1–7.
- [198] C. Liers, M. Pecyna, H. Kellner, A. Worrlich, H. Zorn, K. Steffen, M. Hofrichter, R. Ullrich, Substrate oxidation by dye-decolorizing peroxidases (DyPs) from wood- and litter-degrading agaricomycetes compared to other fungal and plant heme-peroxidases, *Appl. Microbiol. Biotechnol.* 97 (2012).
- [199] C. Chen, T. Li, Bacterial dye-decolorizing peroxidases: biochemical properties and biotechnological opportunities, *Phys. Sci. Rev.* 1 (9) (2016).
- [200] U.A. Zitare, M.H. Habib, H. Rozeboom, M.L. Mascotti, S. Todorovic, M.W. Fraaije, Mutational and structural analysis of an ancestral fungal dye-decolorizing peroxidase, *FEBS J.* 288 (11) (2021) 3602–3618.
- [201] A. Musengi, K. Durrell, A. Prins, N. Khan, M. Agunbiade, T. Kudanga, B. Kirby-McCullough, B.I. Pletschke, S.G. Burton, M. Le Roese-Hill, Production and characterisation of a novel actinobacterial DyP-type peroxidase and its application in coupling of phenolic monomers, *Enzym. Microb. Technol.* 141 (2020), 109654.
- [202] K. Min, G. Gong, H.M. Woo, Y. Kim, Y. Um, A dye-decolorizing peroxidase from *Bacillus subtilis* exhibiting substrate-dependent optimum temperature for dyes and β-ether lignin dimer, *Sci. Rep.* 5 (1) (2015) 8245.
- [203] L. Xu, J. Sun, M.A. Qaria, L. Gao, D. Zhu, Dye decoloring peroxidase structure, catalytic properties and applications: current advancement and futurity, *Catalysts* 11 (8) (2021).
- [204] D. Linde, I. Ayuso-Fernández, M. Laloux, J.E. Aguiar-Cervera, A.L. de Lacey, F. J. Ruiz-Dueñas, A.T. Martínez, Comparing ligninolytic capabilities of bacterial and fungal dye-decolorizing peroxidases and class-II peroxidase-catalases, *Int. J. Mol. Sci.* 22 (5) (2021).
- [205] T. Yoshida, H. Tsuge, H. Konno, T. Hisabori, Y. Sugano, The catalytic mechanism of dye-decolorizing peroxidase DyP may require the swinging movement of an aspartic acid residue, *FEBS J.* 278 (13) (2011) 2387–2394.
- [206] P. Dhankhar, V. Dalal, N. Singh, B.R. Gurjar, A.K. Sharma, P. Kumar, 18 - bioremediation of synthetic dyes: dye decolorizing peroxidases (DyPs), in: M. P. Shah (Ed.), *Removal of Toxic Pollutants Through Microbiological and Tertiary Treatment*, Elsevier, 2020, pp. 453–486.
- [207] L. Xu, J. Sun, M.A. Qaria, L. Gao, D. Zhu, Dye decoloring peroxidase structure, catalytic properties and applications: current advancement and futurity, *Catalysts* (2021).
- [208] M. Adamo, S. Comtet-Marre, E. Büttner, H. Kellner, P. Luis, L. Vallon, R. Prego, M. Hofrichter, M. Giralda, P. Peyret, R. Marmesse, Fungal dye-decolorizing

- peroxidase diversity: roles in either intra- or extracellular processes, *Appl. Microbiol. Biotechnol.* 106 (8) (2022) 2993–3007.
- [209] P. Dhankhar, V. Dalal, A.K. Sharma, P. Kumar, Structural insights at acidic pH of dye-decolorizing peroxidase from *Bacillus subtilis*, *Proteins Struct. Funct. Bioinformatics* 91 (4) (2023) 508–517.
- [210] A. Santos, S. Mendes, V. Brissos, L.O. Martins, New dye-decolorizing peroxidases from *Bacillus subtilis* and *Pseudomonas putida* MET94: towards biotechnological applications, *Appl. Microbiol. Biotechnol.* 98 (5) (2014) 2053–2065.
- [211] Y. Sugano, T. Yoshida, DyP-type peroxidases: recent advances and perspectives, *Int. J. Mol. Sci.* 22 (11) (2021).
- [212] J. Liu, S. Zhang, Q. Shi, L. Wang, W. Kong, H. Yu, F. Ma, Highly efficient oxidation of synthetic and natural lignin-related compounds by *Physporinus vitreus* versatile peroxidase, *Int. Biodeterior. Biodegradation* 136 (2019) 41–48.
- [213] D. Gonzalez-Perez, M. Alcalde, The making of versatile peroxidase by directed evolution, *Biocatalysis Biotransformation* 36 (1) (2018) 1–11.
- [214] D. Gonzalez-Perez, I. Mateljak, E. Garcia-Ruiz, F.J. Ruiz-Dueñas, A.T. Martinez, M. Alcalde, Alkaline versatile peroxidase by directed evolution, *Catal. Sci. Technol.* 6 (17) (2016) 6625–6636.
- [215] S. Barber-Zucker, V. Mindel, E. Garcia-Ruiz, J.J. Weinstein, M. Alcalde, S. J. Fleishman, Stable and functionally diverse versatile peroxidases designed directly from sequences, *J. Am. Chem. Soc.* 144 (8) (2022) 3564–3571.
- [216] N. Rai, M. Yadav, H.S. Yadav, Purification and characterization of versatile peroxidase from citrus sinensis leaf extract and its application in green chemistry, *Anal. Chem. Lett.* 10 (4) (2020) 524–536.
- [217] W. Kong, X. Fu, L. Wang, A. Alhujaili, J. Zhang, F. Ma, X. Zhang, H. Yu, A novel and efficient fungal delignification strategy based on versatile peroxidase for lignocellulose bioconversion, *Biotechnol. Biofuels* 10 (1) (2017) 218.
- [218] J. Zeng, M.J.L. Mills, B.A. Simmons, M.S. Kent, K.L. Sale, Understanding factors controlling depolymerization and polymerization in catalytic degradation of  $\beta$ -ether linked model lignin compounds by versatile peroxidase, *Green Chem.* 19 (9) (2017) 2145–2154.
- [219] X. Wang, B. Yao, X. Su, Linking enzymatic oxidative degradation of lignin to organics detoxification, *Int. J. Mol. Sci.* (2018).
- [220] PDB ID:2BOQ. <https://www.rcsb.org/structure/2BOQ>.
- [221] M. Pérez-Boada, F.J. Ruiz-Dueñas, R. Pogni, R. Basosi, T. Choinowski, M. J. Martínez, K. Piontek, A.T. Martínez, Versatile peroxidase oxidation of high redox potential aromatic compounds: site-directed mutagenesis, spectroscopic and crystallographic investigation of three long-range electron transfer pathways, *J. Mol. Biol.* 354 (2) (2005) 385–402.
- [222] PDB ID:3Q3U. <https://www.rcsb.org/structure/3Q3U>.
- [223] C. Geourjon, G. Deléage, SOPMA: significant improvements in protein secondary structure prediction by consensus prediction from multiple alignments, *Comput. Appl. Biosci.* 11 (6) (1995) 681–684.
- [224] PDB ID:3CG8. <https://www.rcsb.org/structure/3CG8>.
- [225] PDB ID:5VJ0. <https://www.rcsb.org/structure/5VJ0>.
- [226] Lignin peroxidase. [https://www.sigmaaldrich.com/IN/en/product/sigma/42603?gclid=EA1aIqobChMI57zoei\\_gIVh4fCCh11FQoGEAAYASAAEgLZQP\\_D\\_BwE&gclid=aw.ds](https://www.sigmaaldrich.com/IN/en/product/sigma/42603?gclid=EA1aIqobChMI57zoei_gIVh4fCCh11FQoGEAAYASAAEgLZQP_D_BwE&gclid=aw.ds). (Accessed 21 May 2023).
- [227] Laccase. [https://www.sigmaaldrich.com/IN/en/product/sigma/38429?gclid=EA1aIqobChMIkdr13iei\\_gIVma-WCh1a7gC\\_EAAYASAAEgGKPD\\_BwE&gclid=aw.ds](https://www.sigmaaldrich.com/IN/en/product/sigma/38429?gclid=EA1aIqobChMIkdr13iei_gIVma-WCh1a7gC_EAAYASAAEgGKPD_BwE&gclid=aw.ds). (Accessed 21 May 2023).
- [228] Manganese peroxidase. [https://www.sigmaaldrich.com/IN/en/product/sigma/93014?gclid=EA1aIqobChMIjJCX5Yei\\_gIViCRgCh2IfwMFEAAYASAAEgLz3P\\_D\\_BwE&gclid=aw.ds](https://www.sigmaaldrich.com/IN/en/product/sigma/93014?gclid=EA1aIqobChMIjJCX5Yei_gIViCRgCh2IfwMFEAAYASAAEgLz3P_D_BwE&gclid=aw.ds). (Accessed 21 May 2023).
- [229] Versatile peroxidase. <https://www.sigmaaldrich.com/IN/en/tech-docs/paper/300815>. (Accessed 21 May 2023).
- [230] S. Lzaod, T. Dutta, Recent advances in the development of oxidoreductase-based biosensors for detection of phenolic antioxidants in food and beverages, *ACS Omega* 7 (51) (2022) 47434–47448.
- [231] E. Mohit, M. Tabarzad, M.A. Faramarzi, Biomedical and pharmaceutical-related applications of laccases, *Curr. Protein Pept. Sci.* 21 (1) (2020) 78–98.
- [232] A.K. Singh, M. Bilal, H.M.N. Iqbal, A.S. Meyer, A. Raj, Bioremediation of lignin derivatives and phenolics in wastewater with lignin modifying enzymes: status, opportunities and challenges, *Sci. Total Environ.* 777 (2021), 145988.
- [233] R. ten Have, I.M. Rietjens, S. Hartmans, H.J. Swarts, J.A. Field, Calculated ionisation potentials determine the oxidation of vanillin precursors by lignin peroxidase, *FEBS Lett.* 430 (3) (1998) 390–392.
- [234] M. Loi, O. Glazunova, T. Fedorova, A.F. Logrieco, G. Mulè, Fungal laccases: the forefront of enzymes for sustainability, *J. Fungi* (2021).
- [235] F.J. Ruiz-Dueñas, Á.T. Martínez, Microbial degradation of lignin: how a bulky recalcitrant polymer is efficiently recycled in nature and how we can take advantage of this, *Microb. Biotechnol.* 2 (2) (2009) 164–177.
- [236] A. Abhishek, A. Dwivedi, N. Tandan, U. Kumar, Comparative bacterial degradation and detoxification of model and kraft lignin from pulp paper wastewater and its metabolites, *Appl. Water Sci.* 7 (2) (2017) 757–767.
- [237] S. Camarero, B. Bockle, M.J. Martinez, A.T. Martinez, Manganese-mediated lignin degradation by *Pleurotus pulmonarius*, *Appl. Environ. Microbiol.* 62 (3) (1996) 1070–1072.
- [238] G. Mtui, Y. Nakamura, Continuous production of lignin-degrading enzymes by *Bjerkandera adusta* immobilized on polyurethane foam, *Biotechnol. Lett.* 24 (21) (2002) 1743–1747.
- [239] S.K. Chaurasia, N.K. Bhardwaj, Biobleaching - an ecofriendly and environmental benign pulp bleaching technique: a review, *J. Carbohydr. Chem.* 38 (2) (2019) 87–108.
- [240] R.M. Damián-Robles, A.J. Castro-Montoya, J. Saucedo-Luna, M.S. Vázquez-Garcidueñas, M. Arredondo-Santoyo, G. Vázquez-Marrufó, Characterization of ligninolytic enzyme production in white-rot wild fungal strains suitable for kraft pulp bleaching, *Biotech* 7 (5) (2017) 319.
- [241] M.E. Arias, M. Arenas, J. Rodríguez, J. Soliveri, A.S. Ball, M. Hernández, Kraft pulp biobleaching and mediated oxidation of a nonphenolic substrate by laccase from *Streptomyces cyaneus* CECT 3335, *Appl. Environ. Microbiol.* 69 (4) (2003) 1953.
- [242] O.D.V. Biko, M. Viljoen-Bloom, W.H. van Zyl, Microbial lignin peroxidases: applications, production challenges and future perspectives, *Enzym. Microb. Technol.* 141 (2020), 109669.
- [243] O. Fearon, S. Kuitunen, K. Ruuttunen, V. Alopaeus, T. Vuorinen, Detailed modeling of kraft pulping chemistry, *Delignification Ind. Eng. Chem. Res.* 59 (29) (2020) 12977–12985.
- [244] M.H. Lahtinen, J. Mikkilä, K.S. Mikkonen, I. Kilpeläinen, Kraft process—formation of secoisolaricresinol structures and incorporation of fatty acids in kraft lignin, *J. Agric. Food Chem.* 69 (21) (2021) 5955–5965.
- [245] S. Wei, K. Liu, X. Ji, T. Wang, R. Wang, Application of enzyme technology in biopulping and biobleaching, *Cellulose* 28 (16) (2021) 10099–10116.
- [246] C.Y. Liew, A. Husaini, H. Hussain, S. Muid, K.C. Liew, H.A. Roslan, Lignin biodegradation and ligninolytic enzyme studies during biopulping of Acacia mangium wood chips by tropical white rot fungi, *World J. Microbiol. Biotechnol.* 27 (6) (2011) 1457–1468.
- [247] R.T. Mendonça, J.F. Jara, V. González, J.P. Elissetche, J. Freer, Evaluation of the white-rot fungi *Ganoderma australe* and *Ceriporiopsis subvermispora* in biotechnological applications, *J. Ind. Microbiol. Biotechnol.* 35 (11) (2008) 1323–1330.
- [248] K.-Y. Kang, J.-S. Sung, D.-Y. Kim, Evaluation of white-rot fungi for biopulping of wood, *Mycobiology* 35 (4) (2007) 205–209.
- [249] A. Singhal, P.K. Jaiswal, I.S. Thakur, Biopulping of bagasse by *Cryptococcus albidus* under partially sterilized conditions, *Int. Biodeterior. Biodegradation* 97 (2015) 143–150.
- [250] R. Martín-Sampedro, Ú. Fillat, D. Ibarra, M.E. Eugenio, Towards the improvement of *Eucalyptus globulus* chemical and mechanical pulping using endophytic fungi, *Int. Biodeterior. Biodegradation* 105 (2015) 120–126.
- [251] B.S. Orozco Colonia, A. Lorenci Woiciechowski, R. Malanski, L.A. Junior Letti, C. R. Soccol, Pulp improvement of oil palm empty fruit bunches associated to solid-state biopulping and biobleaching with xylanase and lignin peroxidase cocktail produced by *Aspergillus* sp. LPB-5, *Bioresour. Technol.* 285 (2019), 121361.
- [252] G. Singh, N. Capalash, K. Kaur, S. Puri, P. Sharma, Chapter 7 - enzymes: applications in pulp and paper industry, in: G.S. Dhillon, S. Kaur (Eds.), *Agro-industrial Wastes as Feedstock for Enzyme Production*, Academic Press, San Diego, 2016, pp. 157–172.
- [253] S. Trupkin, L. Levin, F. Forchiassin, A. Viale, Optimization of a culture medium for ligninolytic enzyme production and synthetic dye decolorization using response surface methodology, *J. Ind. Microbiol. Biotechnol.* 30 (12) (2003) 682–690.
- [254] D.A. Yaseen, M. Scholz, Textile dye wastewater characteristics and constituents of synthetic effluents: a critical review, *Int. J. Environ. Sci. Technol.* 16 (2) (2019) 1193–1226.
- [255] B. Lellis, C.Z. Fávoro-Polonio, J.A. Pamphile, J.C. Polonio, Effects of textile dyes on health and the environment and bioremediation potential of living organisms, *Biotechnol. Res. Innov.* 3 (2) (2019) 275–290.
- [256] S. Wong, N.A.N. Yac'cob, N. Ngadi, O. Hassan, I.M. Inuwa, From pollutant to solution of wastewater pollution: synthesis of activated carbon from textile sludge for dye adsorption, *Chin. J. Chem. Eng.* 26 (4) (2018) 870–878.
- [257] E. GilPavas, I. Dobrosz-Gómez, M.-Á. Gómez-García, Optimization and toxicity assessment of a combined electrocoagulation, H<sub>2</sub>O<sub>2</sub>/Fe<sup>2+</sup>/UV and activated carbon adsorption for textile wastewater treatment, *Sci. Total Environ.* 651 (2019) 551–560.
- [258] Z. Hussain, M. Arslan, M.H. Malik, M. Mohsin, S. Iqbal, M. Afzal, Treatment of the textile industry effluent in a pilot-scale vertical flow constructed wetland system augmented with bacterial endophytes, *Sci. Total Environ.* 645 (2018) 966–973.
- [259] Z. Hussain, M. Arslan, M.H. Malik, M. Mohsin, S. Iqbal, M. Afzal, Integrated perspectives on the use of bacterial endophytes in horizontal flow constructed wetlands for the treatment of liquid textile effluent: phytoremediation advances in the field, *J. Environ. Manag.* 224 (2018) 387–395.
- [260] H. Yin, P. Qiu, Y. Qian, Z. Kong, X. Zheng, Z. Tang, H. Guo, Textile wastewater treatment for water reuse: a case study, *Processes* 7 (1) (2019).
- [261] G.K. Parshetti, S. Chowdhury, R. Balasubramanian, Hydrothermal conversion of urban food waste to chars for removal of textile dyes from contaminated waters, *Bioresour. Technol.* 161 (2014) 310–319.
- [262] A.A. Telke, S.-W. Kim, S.P. Govindwar, Significant reduction in toxicity, BOD, and COD of textile dyes and textile industry effluent by a novel bacterium *Pseudomonas* sp. LBC1, *Folia Microbiol.* 57 (2) (2012) 115–122.
- [263] J. Fors, M.V. Lindh, J. Pinhassi, U. Welander, Microbial biotreatment of actual textile wastewater in a continuous sequential rice husk biofilter and the microbial community involved, *PLoS One* 12 (1) (2017), e0170562.
- [264] P.D. Kunjadia, G.V. Sanghvi, A.P. Kunjadia, P.N. Mukhopadhyay, G.S. Dave, Role of ligninolytic enzymes of white rot fungi (*Pleurotus* spp.) grown with azo dyes, *SpringerPlus* 5 (1) (2016) 1487.
- [265] K. Vikrant, B.S. Giri, N. Raza, K. Roy, K.-H. Kim, B.N. Rai, R.S. Singh, Recent advancements in bioremediation of dye: current status and challenges, *Bioresour. Technol.* 253 (2018) 355–367.
- [266] G. Parshetti, S. Parshetti, D. Kalyani, R.-a. Doong, S. Govindwar, Industrial dye decolorizing lignin peroxidase from *Kocuria rosea* MTCC 1532, *Ann. Microbiol.* 62 (2011) 217–223.



- [267] W. Tang, X. Xu, B.-C. Ye, P. Cao, A. Ali, Decolorization and degradation analysis of Disperse Red 3B by a consortium of the fungus *Aspergillus* sp. XJ-2 and the microalgae *Chlorella sorokiniana* XJK, RSC Adv. 9 (25) (2019) 14558–14566.
- [268] C.D. Fernandes, V.R.S. Nascimento, D.B. Meneses, D.S. Vilar, N.H. Torres, M. S. Leite, J.R. Vega Baudrit, M. Bilal, H.M.N. Iqbal, R.N. Bharagava, S.M. Egues, L. F. Romanholo Ferreira, Fungal biosynthesis of lignin-modifying enzymes from pulp wash and Luffa cylindrica for azo dye RB5 biodecolorization using modeling by response surface methodology and artificial neural network, J. Hazard. Mater. 399 (2020), 123094.
- [269] Mandeep, G.K. Gupta, H. Liu, P. Shukla, Pulp and paper industry-based pollutants, their health hazards and environmental risks, Curr. Opin. Environ. Sci. Health 12 (2019) 48–56.
- [270] F. Deeba, V. Pruthi, Y.S. Negi, Effect of emerging contaminants from paper mill industry into the environment and their control, in: T. Gupta, A.K. Agarwal, R. A. Agarwal, N.K. Labhsetwar (Eds.), Environmental Contaminants: Measurement, Modelling and Control, Springer Singapore, Singapore, 2018, pp. 391–408.
- [271] C. Ram, P. Rani, K.A. Gebru, M.G. Mariam Abirha, Pulp and paper industry wastewater treatment: use of microbes and their enzymes, Phys. Sci. Rev. 5 (10) (2020).
- [272] J. Ahmed, A. Thakur, A. Goyal, Chapter 1 industrial wastewater and its toxic effects, in: Biological Treatment of Industrial Wastewater, The Royal Society of Chemistry, 2022, pp. 1–14.
- [273] S. Costa, D.G. Dedola, S. Pellizzari, R. Blo, I. Rugiero, P. Pedrini, E. Tamburini, Lignin biodegradation in pulp-and-paper mill wastewater by selected white rot fungi, Water 9 (120) (2017) 935.
- [274] D.D. Mandal, G. Singh, S. Majumdar, P. Chanda, Challenges in developing strategies for the valorization of lignin—a major pollutant of the paper mill industry, Environ. Sci. Pollut. Res. 30 (5) (2023) 11119–11140.
- [275] C. Zhang, F. Wang, Chapter six - catalytic cleavage of lignin CO and CC bonds, in: P.C. Ford, R. van Eldik (Eds.), Advances in Inorganic Chemistry, Academic Press, 2021, pp. 175–218.
- [276] G. Magallanes, M.D. Kärkäs, I. Bosque, S. Lee, S. Maldonado, C.R.J. Stephenson, Selective C–O bond cleavage of lignin systems and polymers enabled by sequential palladium-catalyzed aerobic oxidation and visible-light photoredox catalysis, ACS Catal. 9 (3) (2019) 2252–2260.
- [277] Y. Zheng, M. Guo, Q. Zhou, H. Liu, Effect of lignin degradation product sinapyl alcohol on laccase catalysis during lignin degradation, Ind. Crop. Prod. 139 (2019), 111544.
- [278] X. Chen, B. He, M. Feng, D. Zhao, J. Sun, Immobilized laccase on magnetic nanoparticles for enhanced lignin model compounds degradation, Chin. J. Chem. Eng. 28 (8) (2020) 2152–2159.
- [279] R. Ferrari, V. Gautier, P. Silar, Chapter three - lignin degradation by ascomycetes, in: M. Morel-Rouhier, R. Sormani (Eds.), Advances in Botanical Research, Academic Press, 2021, pp. 77–113.
- [280] L. Gu, M. Wang, H. Li, K.N. Teh, Y. Li, Y. Liu, Tunable Boc modification of lignin and its impact on microbial degradation rate, Sustain. Chem. Pharm. 22 (2021), 100455.
- [281] A. Raychaudhuri, M. Behera, Chapter 9 - microbial degradation of lignin: conversion, application, and challenges, in: M.P. Shah, S. Rodriguez-Couto, R. T. Kapoor (Eds.), Development in Wastewater Treatment Research and Processes, Elsevier, 2022, pp. 195–219.
- [282] A. Ahlawat, S. Mishra, Structural, phylogenetic and expression analysis of genes producing lignin degrading enzymes in the basidiomycete *Cyathus bulleri* and their effect on the release of reducing sugars from agro-residues, Fuel 314 (2022), 122716.
- [283] A. Elframawy, A. El-Hanafy, M. Sharamant, H. Ghozlan, Molecular identification of native Egyptian Actinobacteria: screening for lignin utilization and degradation of lignin model compounds, Biocatal. Agric. Biotechnol. 40 (2022), 102289.
- [284] M. Dou, H. Li, X. Qin, Y. Chang, Y. Wang, C. Zhao, Z. Liu, C. Gu, Y. Fu, An efficient strategy to degrade lignin by a novel endophytic fungus *Chaetomium globosum* combined with ultrasound, Ind. Crop. Prod. 178 (2022), 114578.
- [285] S.I. Khan, N.S. Zada, M. Sahinkaya, D. Nigar Colak, S. Ahmed, F. Hasan, A. O. Belduz, S. Çanakçı, S. Khan, M. Badshah, A.A. Shah, Cloning, expression and biochemical characterization of lignin-degrading DyP-type peroxidase from *Bacillus* sp. strain BL5, Enzym. Microb. Technol. 151 (2021), 109917.
- [286] K. Murugesan, Bioremediation of paper and pulp mill effluents, Indian J. Exp. Biol. 41 (11) (2003) 1239–1248.
- [287] J. Duan, X. Huo, W.J. Du, J.D. Liang, D.Q. Wang, S.C. Yang, Biodegradation of kraft lignin by a newly isolated anaerobic bacterial strain, *Acetanaerobium* sp. WJDL-Y2, Lett. Appl. Microbiol. 62 (1) (2016) 55–62.
- [288] A.C. Freitas, F. Ferreira, A.M. Costa, R. Pereira, S.C. Antunes, F. Gonçalves, T. A. Rocha-Santos, M.S. Diniz, L. Castro, I. Peres, A.C. Duarte, Biological treatment of the effluent from a bleached kraft pulp mill using basidiomycete and zygomycete fungi, Sci. Total Environ. 407 (10) (2009) 3282–3289.
- [289] A.K. Singh, P. Yadav, R.N. Bharagava, G.D. Saratale, A. Raj, Biotransformation and cytotoxicity evaluation of kraft lignin degraded by ligninolytic *Serratia liquefaciens* 10 (2019) 2364.
- [290] K. Deng, J. Zeng, G. Cheng, J. Gao, K.L. Sale, B.A. Simmons, A.K. Singh, P. D. Adams, T.R. Northen, Rapid characterization of the activities of lignin-modifying enzymes based on nanostructure-initiator mass spectrometry (NIMS), Biotechnol. Biofuels 11 (1) (2018) 266.
- [291] V. Christian, R. Shrivastava, D. Shukla, H. Modi, B. Vyas, Degradation of xenobiotic compounds by lignin-degrading white-rot fungi: enzymology and mechanisms involved, Indian J. Exp. Biol. 43 (2005) 301–312.
- [292] N. Agrawal, P. Verma, S.K. Shahi, Degradation of polycyclic aromatic hydrocarbons (phenanthrene and pyrene) by the ligninolytic fungi *Ganoderma lucidum* isolated from the hardwood stump, Bioresour. Bioprocess. 5 (1) (2018) 1–9.
- [293] M. Cameron, S. Timofeevski, S. Aust, Enzymology of *Phanerochaete chrysosporium* with respect to the degradation of recalcitrant compounds and xenobiotics, Appl. Microbiol. Biotechnol. 54 (6) (2000) 751–758.
- [294] T. Cui, B. Yuan, H. Guo, H. Tian, W. Wang, Y. Ma, C. Li, Q. Fei, Enhanced lignin biodegradation by consortium of white rot fungi: microbial synergistic effects and product mapping, Biotechnol. Biofuels 14 (1) (2021) 162.
- [295] D. Salvachúa, R. Katahira, N.S. Cleveland, P. Khanna, M.G. Resch, B.A. Black, S. O. Purvine, E.M. Zink, A. Prieto, M.J. Martínez, A.T. Martínez, B.A. Simmons, J. M. Gladden, G.T. Beckham, Lignin depolymerization by fungal secretomes and a microbial sink, Green Chem. 18 (22) (2016) 6046–6062.
- [296] C.C. Gonçalves, T. Bruce, C.d.O.G. Silva, E.X.F. Filho, E.F. Noronha, M. Carlquist, N.S. Parachin, Bioprospecting microbial diversity for lignin valorization: dry and wet screening methods, Front. Microbiol. 11 (2020).
- [297] J. Plácido, S. Capareda, Ligninolytic enzymes: a biotechnological alternative for bioethanol production, Bioresour. Bioprocess. 2 (1) (2015) 23.
- [298] N. Ahmad, S. Aslam, N. Hussain, M. Bilal, H.M.N. Iqbal, Transforming lignin biomass to value: interplay between ligninolytic enzymes and lignocellulose depolymerization, BioEnergy Res. (2022).
- [299] E. Rosini, F. Molinari, D. Miani, L. Pollegioni, Lignin valorization: production of high value-added compounds by engineered microorganisms, Catalysts (2023).
- [300] L. Yang, K. Seshan, Y. Li, A review on thermal chemical reactions of lignin model compounds, Catal. Today 298 (2017) 276–297.
- [301] T. Elder, J.J. Bozell, D. Cedenio, The effect of axial ligand on the oxidation of syringyl alcohol by Co(salen) adducts, Phys. Chem. Chem. Phys. 15 (19) (2013) 7328–7337.
- [302] D. Varhanickova, S.C. Lee, W.-Y. Shiu, D. Mackay, Aqueous solubilities of chlorocatechols, chlorovanillins, chlorosyringols, and chlorosyringaldehydes at 25 °C, J. Chem. Eng. Data 40 (3) (1995) 620–622.
- [303] V.S. Haritos, C.M. Brumley, D.A. Holdway, J.T. Ahokas, Metabolites of 2-chlorosyringaldehyde in fish bile: indicator of exposure to bleached hardwood effluent, Xenobiotica 25 (9) (1995) 963–971.
- [304] S.C. Patankar, L.-Y. Liu, L. Ji, S. Ayakar, V. Yadav, S. Rennecker, Isolation of phenolic monomers from kraft lignin using a magnetically recyclable TEMPO nanocatalyst, Green Chem. 21 (4) (2019) 785–791.
- [305] J.C. del Río, J. Rencoret, A. Gutiérrez, T. Elder, H. Kim, J. Ralph, Lignin monomers from beyond the canonical monolignol biosynthetic pathway: another brick in the wall, ACS Sustain. Chem. Eng. 8 (13) (2020) 4997–5012.
- [306] G. Liu, Y. Zhao, J. Guo, High selectively catalytic conversion of lignin-based phenols into para-/m-Xylene over Pt/HZSM-5, Catalysts (2016).
- [307] I.B.D. de Castro, I. Graça, L. Rodríguez-García, M. Kennema, R. Rinaldi, F. Meemken, Elucidating the reactivity of methoxyphenol positional isomers towards hydrogen-transfer reactions by ATR-IR spectroscopy of the liquid–solid interface of RANEY® Ni, Catal. Sci. Technol. 8 (12) (2018) 3107–3114.
- [308] J. Zhang, L. Lombardo, G. Gözyaydin, P.J. Dyson, N. Yan, Single-step conversion of lignin monomers to phenol: bridging the gap between lignin and high-value chemicals, Chin. J. Catal. 39 (9) (2018) 1445–1452.
- [309] X. Chen, W. Guan, C.-W. Tsang, H. Hu, C. Liang, Lignin valorizations with Ni catalysts for renewable chemicals and fuels productions, Catalysts 9 (6) (2019).
- [310] R. Margesin, G. Volgger, A.O. Wagner, D. Zhang, C. Poyntner, Biodegradation of lignin monomers and bioconversion of ferulic acid to vanillic acid by *Paraburkholderia aromaticivorans* AR20-38 isolated from Alpine forest soil, Appl. Microbiol. Biotechnol. 105 (7) (2021) 2967–2977.
- [311] C. Cai, Z. Xu, H. Zhou, S. Chen, M. Jin, Valorization of lignin components into gallate by integrated biological hydroxylation, O-demethylation, and aryl side-chain oxidation, Sci. Adv. 7(36) eabg4585.
- [312] H. Kim, Q. Li, S.D. Karlen, R.A. Smith, R. Shi, J. Liu, C. Yang, S. Tunlaya-Anukit, J. P. Wang, H.-M. Chang, R.R. Sederoff, J. Ralph, V.L. Chiang, Monolignol benzoates incorporate into the lignin of transgenic *Populus trichocarpa* depleted in C3H and C4H, ACS Sustain. Chem. Eng. 8 (9) (2020) 3644–3654.
- [313] K. Ravi, J. García-Hidalgo, M.F. Gorwa-Grauslund, G. Lidén, Conversion of lignin model compounds by *Pseudomonas putida* KT2440 and isolates from compost, Appl. Microbiol. Biotechnol. 101 (12) (2017) 5059–5070.
- [314] G. Ward, Y. Hadar, I. Bilkis, L. Konstantinovskiy, C.G. Dosoretz, Initial steps of ferulic acid polymerization by lignin peroxidase, J. Biol. Chem. 276 (22) (2001) 18734–18741.
- [315] D. Jo Heuschele, K.P. Smith, G.A. Annor, Variation in lignin, cell wall-bound p-coumaric, and ferulic acid in the nodes and internodes of cereals and their impact on lodging, J. Agric. Food Chem. 68 (45) (2020) 12569–12576.
- [316] R. Panyadee, A. Saengsrirachan, P. Posoknistakul, N. Laosiripojana, S. Ratchahat, B. M. Matsagar, K.C.W. Wu, C. Sakdaronnarong, Lignin-derived syringol and acetosyringone from palm bunch using heterogenous oxidative depolymerization over mixed metal oxide catalysts under microwave heating, Molecules 26 (24) (2021).
- [317] J. Rönnander, J. Ljunggren, E. Hedenström, S.A.I. Wright, Biotransformation of vanillin into vanillyl alcohol by a novel strain of *Cystobasidium laryngis* isolated from decaying wood, AMB Express 8 (1) (2018) 137.
- [318] J. Yang, T. Gao, Y. Zhang, S. Wang, H. Li, S. Li, S. Wang, Degradation of the phenolic  $\beta$ -ether lignin model dimer and dyes by dye-decolorizing peroxidase from *Bacillus amyloliquefaciens*, Biotechnol. Lett. 41 (8–9) (2019) 1015–1021.
- [319] Y. Liu, G. Lyu, X. Ji, G. Yang, J. Chen, L.A. Lucia, Analytical pyrolysis pathways of guaiacyl glycerol- $\beta$ -guaiacyl ether by Py-GC/MS, BioResources 11 (3) (2016).
- [320] M. Awasthi, N. Jaiswal, S. Singh, V.P. Pandey, U.N. Dwivedi, Molecular docking and dynamics simulation analyses unraveling the differential enzymatic catalysis

- by plant and fungal laccases with respect to lignin biosynthesis and degradation, *J. Biomol. Struct. Dyn.* 33 (9) (2015) 1835–1849.
- [321] Y. Shi, X. Yan, Q. Li, X. Wang, S. Xie, L. Chai, J. Yuan, Directed bioconversion of Kraft lignin to polyhydroxyalkanoate by *Cupriavidus basilensis* B-8 without any pretreatment, *Process Biochem.* 52 (2017) 238–242.
- [322] M. Si, X. Yan, M. Liu, M. Shi, Z. Wang, S. Wang, J. Zhang, C. Gao, L. Chai, Y. Shi, In situ lignin bioconversion promotes complete carbohydrate conversion of rice straw by *Cupriavidus basilensis* B-8, *ACS Sustain. Chem. Eng.* 6 (6) (2018) 7969–7978.
- [323] Y. Yang, W.Y. Song, H.G. Hur, T.Y. Kim, S. Ghatge, Thermoalkaliphilic laccase treatment for enhanced production of high-value benzaldehyde chemicals from lignin, *Int. J. Biol. Macromol.* 124 (2019) 200–208.
- [324] K. Saikia, D. Vishnu, A.K. Rathankumar, B. Palanisamy Athiyaman, R.A. Batista-García, J.L. Folch-Mallol, H. Cabana, V.V. Kumar, Development of a magnetically separable co-immobilized laccase and versatile peroxidase system for the conversion of lignocellulosic biomass to vanillin, *J. Air Waste Manag. Assoc.* (1995) 70 (12) (2020) 1252–1259.
- [325] M.B. Agustin, D.M. de Carvalho, M.H. Lahtinen, K. Hilden, T. Lundell, K. S. Mikkonen, Laccase as a tool in building advanced lignin-based materials, *ChemSusChem* 14 (21) (2021) 4615–4635.
- [326] Z. Xu, P. Lei, R. Zhai, Z. Wen, M. Jin, Recent advances in lignin valorization with bacterial cultures: microorganisms, metabolic pathways, and bio-products, *Biotechnol. Biofuels* 12 (1) (2019) 32.
- [327] C.W. Johnson, G.T. Beckham, Aromatic catabolic pathway selection for optimal production of pyruvate and lactate from lignin, *Metab. Eng.* 28 (2015) 240–247.
- [328] M. Kumar, A. Singhal, P.K. Verma, I.S. Thakur, Production and characterization of polyhydroxyalkanoate from lignin derivatives by *Pandora sp.* ISTKB, *ACS Omega* 2 (12) (2017) 9156–9163.
- [329] C.-Y. Hong, S.-H. Ryu, H. Jeong, S.-S. Lee, M. Kim, I.-G. Choi, *Phanerochaete chrysosporium* multienzyme catabolic system for in vivo modification of synthetic lignin to succinic acid, *ACS Chem. Biol.* 12 (7) (2017) 1749–1759.
- [330] T. Sonoki, K. Takahashi, H. Sugita, M. Hatamura, Y. Azuma, T. Sato, S. Suzuki, N. Kamimura, E. Masai, Glucose-free cis, cis-muconic acid production via new metabolic designs corresponding to the heterogeneity of lignin, *ACS Sustain. Chem. Eng.* 6 (1) (2018) 1256–1264.
- [331] D.R. Vardon, M.A. Franden, C.W. Johnson, E.M. Karp, M.T. Guarnieri, J.G. Linger, M.J. Salm, T.J. Strathmann, G.T. Beckham, Adipic acid production from lignin, *Energy Environ. Sci.* 8 (2) (2015) 617–628.
- [332] Z.-H. Liu, S. Xie, F. Lin, M. Jin, J.S. Yuan, Combinatorial pretreatment and fermentation optimization enable a record yield on lignin bioconversion, *Biotechnol. Biofuels* 11 (1) (2018) 1–20.
- [333] C. Zhao, S. Xie, Y. Pu, R. Zhang, F. Huang, A.J. Ragauskas, J.S. Yuan, Synergistic enzymatic and microbial lignin conversion, *Green Chem.* 18 (5) (2016) 1306–1312.
- [334] S.R. Couto, J.L.T. Herrera, Industrial and biotechnological applications of laccases: a review, *Biotechnol. Adv.* 24 (5) (2006) 500–513.
- [335] P. Chen, W. Chen, S. Jiang, Q. Zhong, H. Chen, W. Chen, Synergistic effect of laccase and sugar beet pectin on the properties of concentrated protein emulsions and its application in concentrated coconut milk, *Molecules* (2018).
- [336] K. Mayolo-Deloiisa, M. González-González, M. Rito-Palomares, Laccases in food industry: bioprocessing, potential industrial and biotechnological applications, *Front. Bioeng. Biotechnol.* 8 (2020) 222.
- [337] J.F. Osma, J.L. Toca-Herrera, S. Rodríguez-Couto, Uses of laccases in the food industry, *Enzyme Res.* 2010 (2010), 918761.
- [338] A. Mookoolall, L. Sykora, J. Pfannstiel, S. Nöbel, J. Weiss, J. Hinrichs, A feasibility study on the application of a laccase-mediator system in stirred yoghurt at the pilot scale, *Food Hydrocoll.* 60 (2016) 119–127.
- [339] C.S.M. Bezerra-Filho, J.N. Barboza, M.T.S. Souza, P. Sabry, N.S.M. Ismail, D.P. de Sousa, Therapeutic potential of vanillin and its main metabolites to regulate the inflammatory response and oxidative stress, *Mini-Rev. Med. Chem.* 19 (20) (2019) 1681–1693.
- [340] ten Have, Lignin Peroxidase Mediated Biotransformations Useful in the Biocatalytic Production of Vanillin, Wageningen University, 2000, ISBN 90-5808-169-9 (Promotor: Prof.dr.ir. J.A.M. de Bont, co-promotor(en): Dr. J.A. Field. - Wageningen : R. ten Have, 2000).
- [341] R. ten Have, I.M.C.M. Rietjens, S. Hartmans, H.J. Swarts, J.A. Field, Calculated ionisation potentials determine the oxidation of vanillin precursors by lignin peroxidase, *FEBS Lett.* 430 (3) (1998) 390–392.
- [342] K. Saikia, D. Vishnu, A.K. Rathankumar, B. Palanisamy Athiyaman, R.A. Batista-García, J.L. Folch-Mallol, H. Cabana, V.V. Kumar, Development of a magnetically separable co-immobilized laccase and versatile peroxidase system for the conversion of lignocellulosic biomass to vanillin, *J. Air Waste Manag. Assoc.* 70 (12) (2020) 1252–1259.
- [343] K. Mayolo-Deloiisa, M. González-González, M. Rito-Palomares, Laccases in food industry: bioprocessing, potential industrial and biotechnological applications, *Front. Bioeng. Biotechnol.* 8 (2020) 222.
- [344] A.D. Moreno, D. Ibarra, M.E. Eugenio, E. Tomás-Pejó, Laccases as versatile enzymes: from industrial uses to novel applications, *J. Chem. Technol. Biotechnol.* 95 (3) (2020) 481–494.
- [345] S. Raveendran, B. Parameswaran, S.B. Ummalyma, A. Abraham, A.K. Mathew, A. Madhavan, S. Rebello, A. Pandey, Applications of microbial enzymes in food industry, *Food Technol. Biotechnol.* 56 (1) (2018) 16–30.
- [346] M. Patel, R. Kumar, K. Kishor, T. Mlsna, C.U. Pittman Jr., D. Mohan, Pharmaceuticals of emerging concern in aquatic systems: chemistry, occurrence, effects, and removal methods, *Chem. Rev.* 119 (6) (2019) 3510–3673.
- [347] W. Xu, R. Zou, B. Jin, G. Zhang, Y. Su, Y. Zhang, The ins and outs of pharmaceutical wastewater treatment by microbial electrochemical technologies, *Sustain. Horiz.* 1 (2022), 100003.
- [348] A. Apriceno, M.L. Astolfi, A.M. Girelli, F.R. Scuto, A new laccase-mediator system facing the biodegradation challenge: insight into the NSAIDs removal, *Chemosphere* 215 (2019) 535–542.
- [349] B. Almeida, A. Oehmen, R. Marques, D. Brito, G. Carvalho, M.T. Crespo, Modelling the biodegradation of non-steroidal anti-inflammatory drugs (NSAIDs) by activated sludge and a pure culture, *Bioresour. Technol.* 133 (2013) 31–37.
- [350] D. Domaradzka, U. Guzik, D. Wojcieszynska, Biodegradation and biotransformation of polycyclic non-steroidal anti-inflammatory drugs, *Rev. Environ. Sci. Biotechnol.* 14 (2) (2015) 229–239.
- [351] D. Wojcieszynska, U. Guzik, Naproxen in the environment: its occurrence, toxicity to nontarget organisms and biodegradation, *Appl. Microbiol. Biotechnol.* 104 (5) (2020) 1849–1857.
- [352] Y.D. Aracagök, H. Göker, N. Cihangir, Biodegradation of micropollutant naproxen with a selected fungal strain and identification of metabolites, *Z. Naturforsch. C* 72 (5–6) (2017) 173–179.
- [353] T.K. Kasonga, I. Kamika, V.M. Ngole-Jeme, Ligninolytic enzyme activity and removal efficiency of pharmaceuticals in a water matrix by fungus *Rhizopus* sp. isolated from cassava, *Environ. Technol.* (2022) 1–14.
- [354] S.K. Alharbi, L.D. Nghiem, J.P. van de Merwe, F.D.L. Leusch, M.B. Asif, F.I. Hai, W.E. Price, Degradation of diclofenac, trimethoprim, carbamazepine, and sulfamethoxazole by laccase from *Trametes versicolor*: transformation products and toxicity of treated effluent, *Biocatalysis Biotransformation* 37 (6) (2019) 399–408.
- [355] M. Meiczinger, B. Varga, L. Wolmarans, L. Hajba, V. Somogyi, Stability improvement of laccase for micropollutant removal of pharmaceutical origins from municipal wastewater, *Clean Techn. Environ. Policy* 24 (10) (2022) 3213–3223.
- [356] K.K. Navada, A. Kulal, Enzymatic degradation of chloramphenicol by laccase from *Trametes hirsuta* and comparison among mediators, *Int. Biodeterior. Biodegradation* 138 (2019) 63–69.
- [357] M. Kelbert, C.S. Pereira, N.A. Daronch, K. Cesca, C. Michels, D. de Oliveira, H. M. Soares, Laccase as an efficacious approach to remove anticancer drugs: a study of doxorubicin degradation, kinetic parameters, and toxicity assessment, *J. Hazard. Mater.* 409 (2021), 124520.
- [358] H. Cabana, J. Jones, S.N. Agathos, Elimination of endocrine disrupting chemicals using white rot fungi and their lignin modifying enzymes: a review, *Eng. Life Sci.* 7 (5) (2007) 429–456.
- [359] EDCs. <https://www.endocrine.org/patient-engagement/endocrine-library/edcs>. (Accessed 21 May 2023).
- [360] EDCs. <https://www.niehs.nih.gov/health/topics/agents/endocrine/index.cfm>. (Accessed 21 May 2023).
- [361] E. Diamanti-Kandarakis, J.-P. Bourguignon, L.C. Giudice, R. Hauser, G.S. Prins, A. M. Soto, R.T. Zoeller, A.C. Gore, Endocrine-disrupting chemicals: an Endocrine Society scientific statement, *Endocr. Rev.* 30 (4) (2009) 293–342.
- [362] W.H. Organization, State of the Science of Endocrine Disrupting Chemicals 2012: Summary for Decision-makers, 2012.
- [363] J. Annamalai, V. Namasivayam, Endocrine disrupting chemicals in the atmosphere: their effects on humans and wildlife, *Environ. Int.* 76 (2015) 78–97.
- [364] J. Annamalai, V. Namasivayam, Endocrine disrupting chemicals in the atmosphere: their effects on humans and wildlife, *Environ. Int.* 76 (2015) 78–97.
- [365] T. Rasheed, M. Bilal, F. Nabeel, M. Adeel, H.M.N. Iqbal, Environmentally-related contaminants of high concern: potential sources and analytical modalities for detection, quantification, and treatment, *Environ. Int.* 122 (2019) 52–66.
- [366] M. Bilal, M. Adeel, T. Rasheed, Y. Zhao, H.M.N. Iqbal, Emerging contaminants of high concern and their enzyme-assisted biodegradation – a review, *Environ. Int.* 124 (2019) 336–353.
- [367] M. Kumar, D.K. Sarma, S. Shubham, M. Kumawat, V. Verma, A. Prakash, R. Tiwari, Environmental endocrine-disrupting chemical exposure: role in non-communicable diseases, *Front. Public Health* 8 (2020).
- [368] M. Bilal, M. Adeel, T. Rasheed, Y. Zhao, H.M.N. Iqbal, Emerging contaminants of high concern and their enzyme-assisted biodegradation - a review, *Environ. Int.* 124 (2019) 336–353.
- [369] D. Becker, S. Rodriguez-Mozaz, S. Insa, R. Schoevaert, D. Barceló, M. de Cazes, M.-P. Belleville, J. Sanchez-Marcano, A. Misovic, J. Oehlmann, M. Wagner, Removal of endocrine disrupting chemicals in wastewater by enzymatic treatment with fungal laccases, *Org. Process. Res. Dev.* 21 (4) (2017) 480–491.
- [370] A. Gałzka, U. Jankiewicz, Endocrine disrupting compounds (nonylphenol and bisphenol A)-sources, harmfulness and laccase-assisted degradation in the aquatic environment, *Microorganisms* (2022).
- [371] A. Grelska, M. Noszczyńska, White rot fungi can be a promising tool for removal of bisphenol A, bisphenol S, and nonylphenol from wastewater, *Environ. Sci. Pollut. Res.* 27 (32) (2020) 39958–39976.
- [372] R. Taboada-Puig, T.A. Lu-Chau, G. Eibes, G. Feijoo, M.T. Moreira, J.M. Lema, Continuous removal of endocrine disruptors by versatile peroxidase using a two-stage system, *Biotechnol. Prog.* 31 (4) (2015) 908–916.
- [373] W. Zheng, L.M. Colosi, Peroxidase-mediated removal of endocrine disrupting compound mixtures from water, *Chemosphere* 85 (4) (2011) 553–557.
- [374] R. Taboada-Puig, G. Eibes, L. Lloret, T.A. Lu-Chau, G. Feijoo, M.T. Moreira, J. M. Lema, Fostering the action of versatile peroxidase as a highly efficient biocatalyst for the removal of endocrine disrupting compounds, *New Biotechnol.* 33 (1) (2016) 187–195.

- [375] J. Wang, N. Majima, H. Hirai, H. Kawagishi, Effective removal of endocrine-disrupting compounds by lignin peroxidase from the white-rot fungus *Phanerochaete sordida* YK-624, *Curr. Microbiol.* 64 (3) (2012) 300–303.
- [376] H. Cabana, J.P. Jones, S.N. Agathos, Elimination of endocrine disrupting chemicals using white rot fungi and their lignin modifying enzymes: a review, *Eng. Life Sci.* 7 (5) (2007) 429–456.
- [377] C. Pezzella, G. Macellaro, G. Sannia, F. Raganati, G. Olivieri, A. Marzocchella, D. Schlosser, A. Piscitelli, Exploitation of *Trametes versicolor* for bioremediation of endocrine disrupting chemicals in bioreactors, *PLoS One* 12 (6) (2017), e0178758.
- [378] K. Sei, T. Takeda, S.O. Soda, M. Fujita, M. Ike, Removal characteristics of endocrine-disrupting chemicals by laccase from white-rot fungi, *J. Environ. Sci. Health A Tox. Hazard. Subst. Environ. Eng.* 43 (1) (2008) 53–60.
- [379] Z. Asadgol, H. Forootanfar, S. Rezaei, A.H. Mahvi, M.A. Faramarzi, Removal of phenol and bisphenol-A catalyzed by laccase in aqueous solution, *J. Environ. Health Sci. Eng.* 12 (1) (2014) 93.
- [380] G. Songulashvili, G.A. Jimenez-Tobón, C. Jaspers, M.J. Penninckx, Immobilized laccase of *Cerrena unicolor* for elimination of endocrine disruptor micropollutants, *Fungal Biol.* 116 (8) (2012) 883–889.
- [381] L. Lloret, G. Eibes, M.T. Moreira, G. Feijoo, J.M. Lema, Removal of estrogenic compounds from filtered secondary wastewater effluent in a continuous enzymatic membrane reactor. Identification of biotransformation products, *Environ. Sci. Technol.* 47 (9) (2013) 4536–4543.
- [382] L. Lloret, G. Eibes, G. Feijoo, M.T. Moreira, J.M. Lema, F. Hollmann, Immobilization of laccase by encapsulation in a sol-gel matrix and its characterization and use for the removal of estrogens, *Biotechnol. Prog.* 27 (6) (2011) 1570–1579.
- [383] M. Bilal, H.M.N. Iqbal, D. Barceló, Mitigation of bisphenol A using an array of laccase-based robust bio-catalytic cues - a review, *Sci. Total Environ.* 689 (2019) 160–177.
- [384] M. Bilal, H.M.N. Iqbal, Persistence and impact of steroidal estrogens on the environment and their laccase-assisted removal, *Sci. Total Environ.* 690 (2019) 447–459.
- [385] M. Bilal, H.M.N. Iqbal, D. Barceló, Persistence of pesticides-based contaminants in the environment and their effective degradation using laccase-assisted biocatalytic systems, *Sci. Total Environ.* 695 (2019), 133896.
- [386] G. Macellaro, C. Pezzella, P. Cicatiello, G. Sannia, A. Piscitelli, Fungal laccases degradation of endocrine disrupting compounds, *Biomed. Res. Int.* 2014 (2014), 614038.
- [387] Y. Dai, J. Yao, Y. Song, X. Liu, S. Wang, Y. Yuan, Enhanced performance of immobilized laccase in electrospun fibrous membranes by carbon nanotubes modification and its application for bisphenol A removal from water, *J. Hazard. Mater.* 317 (2016) 485–493.
- [388] J. Zdzarta, K. Anteck, R. Frankowski, A. Zgola-Grzeskowiak, H. Ehrlich, T. Jesionowski, The effect of operational parameters on the biodegradation of bisphenols by *Trametes versicolor* laccase immobilized on Hippopongia communis spongin scaffolds, *Sci. Total Environ.* 615 (2018) 784–795.
- [389] R. Taboada-Puig, T.A. Lu-Chau, G. Eibes, G. Feijoo, M.T. Moreira, J.M. Lema, Continuous removal of endocrine disruptors by versatile peroxidase using a two-stage system, *Biotechnol. Prog.* 31 (4) (2015) 908–916.
- [390] K. Chen, S. Liu, Q. Zhang, Degradation and detection of endocrine disruptors by laccase-mimetic polyoxometalates, *Front. Chem.* 10 (2022), 854045.
- [391] S. Sarkar, A. Banerjee, U. Halder, R. Biswas, R. Bandopadhyay, Degradation of synthetic azo dyes of textile industry: a sustainable approach using microbial enzymes, *Water Conserv. Sci. Eng.* 2 (4) (2017) 121–131.
- [392] P. Sathishkumar, F.L. Gu, F. Ameen, T. Palvannan, Fungal laccase mediated bioremediation of xenobiotic compounds, in: *Microbial Biodegradation of Xenobiotic Compounds*, CRC Press, 2019, pp. 135–157.
- [393] E. Balcázar-López, L.H. Méndez-Lorenzo, R.A. Batista-García, U. Esquivel-Naranjo, M. Ayala, V.V. Kumar, O. Savary, H. Cabana, A. Herrera-Estrella, J. L. Folch-Mallol, Xenobiotic compounds degradation by heterologous expression of a *Trametes sanguineus* laccase in *Trichoderma atroviride*, *PLoS One* 11 (2) (2016), e0147997.
- [394] M. Bilal, A.R. Bagheri, D.S. Vilar, N. Aramesh, K.I.B. Eguiluz, L.F.R. Ferreira, S. S. Ashraf, H.M.N. Iqbal, Oxidoreductases as a versatile biocatalytic tool to tackle pollutants for clean environment – a review, *J. Chem. Technol. Biotechnol.* 97 (2) (2022) 420–435.
- [395] M. Bilal, S.S. Ashraf, D. Barceló, H.M.N. Iqbal, Biocatalytic degradation/ redefining “removal” fate of pharmaceutically active compounds and antibiotics in the aquatic environment, *Sci. Total Environ.* 691 (2019) 1190–1211.
- [396] P.K. Chaurasia, S.L. Bharati, C. Sarma, Laccases in pharmaceutical chemistry: a comprehensive appraisal, *Mini-Rev. Org. Chem.* 13 (6) (2016) 430–451.
- [397] H.J. Sung, M.F. Khan, Y.H. Kim, Recombinant lignin peroxidase-catalyzed decolorization of melanin using in-situ generated H<sub>2</sub>O<sub>2</sub> for application in whitening cosmetics, *Int. J. Biol. Macromol.* 136 (2019) 20–26.
- [398] B. Sadaqat, N. Khatoun, A.Y. Malik, A. Jamal, U. Farooq, M.I. Ali, H. He, F.-J. Liu, H. Guo, M. Urynowicz, Q. Wang, Z. Huang, Enzymatic decolorization of melanin by lignin peroxidase from *Phanerochaete chrysosporium*, *Sci. Rep.* 10 (1) (2020) 20240.
- [399] S.-M. Zhong, N. Sun, H.-X. Liu, Y.-Q. Niu, Y. Wu, Reduction of facial pigmentation of melasma by topical lignin peroxidase: a novel fast-acting skin-lightening agent, *Exp. Ther. Med.* 9 (2) (2015) 341–344.
- [400] J. Baik, A. Purkayastha, K.H. Park, T.J. Kang, Functional characterization of melanin decolorizing extracellular peroxidase of *Bjerkandera adusta*, *J. Fungi* (Basel, Switzerland) 7 (9) (2021).
- [401] H.J. Sung, M.F. Khan, Y.H. Kim, Recombinant lignin peroxidase-catalyzed decolorization of melanin using in-situ generated H<sub>2</sub>O<sub>2</sub> for application in whitening cosmetics, *Int. J. Biol. Macromol.* 136 (2019) 20–26.
- [402] L. Liu, S.A. Qamar, M. Bilal, H.M.N. Iqbal, Broadening the catalytic role of enzymes in cosmeceutical sector: a robust tool from white biotechnology, *Catal. Lett.* 152 (2022) 707+.
- [403] S.M. Zhong, N. Sun, H.X. Liu, Y.Q. Niu, Y. Wu, Reduction of facial pigmentation of melasma by topical lignin peroxidase: a novel fast-acting skin-lightening agent, *Exp. Ther. Med.* 9 (2) (2015) 341–344.
- [404] S.K. Shin, J.E. Hyeon, Y.C. Joo, D.W. Jeong, S.K. You, S.O. Han, Effective melanin degradation by a synergistic laccase-peroxidase enzyme complex for skin whitening and other practical applications, *Int. J. Biol. Macromol.* 129 (2019) 181–186.
- [405] A. Zerva, S. Simić, E. Topakas, J. Nikodinovic-Runic, Applications of microbial laccases: patent review of the past decade (2009–2019), *Catalysts* 9 (12) (2019).
- [406] N. Cardullo, L. Pulvirenti, C. Spatafora, N. Musso, V. Barresi, D.F. Condorelli, C. Tringali, Dihydrobenzofuran neolignanamide: laccase-mediated biomimetic synthesis and antiproliferative activity, *J. Nat. Prod.* 79 (8) (2016) 2122–2134.
- [407] K.W. Wellington, R. Bokako, N. Raseroka, P. Steenkamp, A one-pot synthesis of 1,4-naphthoquinone-2,3-bis-sulfides catalysed by a commercial laccase, *Green Chem.* 14 (9) (2012) 2567–2576.
- [408] K.W. Wellington, N.I. Kolesnikova, V. Hlatshwayo, S.T. Saha, M. Kaur, L. R. Motadi, Anticancer activity, apoptosis and a structure–activity analysis of a series of 1, 4-naphthoquinone-2, 3-bis-sulfides, *Investig. New Drugs* 38 (2) (2020) 274–286.
- [409] D. Kumar, A. Kumar, S. Sondhi, P. Sharma, N. Gupta, An alkaline bacterial laccase for polymerization of natural precursors for hair dye synthesis, *3, Biotech* 8 (3) (2018) 182.
- [410] O.J. Morel, R.M. Christie, Current trends in the chemistry of permanent hair dyeing, *Chem. Rev.* 111 (4) (2011) 2537–2561.
- [411] K. Otsuka Saito, R. Ikeda, K. Endo, Y. Tsujino, M. Takagi, E. Tamiya, Isolation of a novel alkaline-induced laccase from *Flammulina velutipes* and its application for hair coloring, *J. Biosci. Bioeng.* 113 (5) (2012) 575–579.
- [412] J. An, G. Li, Y. Zhang, T. Zhang, X. Liu, F. Gao, M. Peng, Y. He, H. Fan, Recent advances in enzyme-nanostructure biocatalysts with enhanced activity, *Catalysts* 10 (3) (2020).
- [413] E. Gkantzou, A.V. Chatzikonstantinou, R. Fotiadou, A. Giannakopoulou, M. Patila, H. Stamatis, Trends in the development of innovative nanobiocatalysts and their application in biocatalytic transformations, *Biotechnol. Adv.* 51 (2021), 107738.
- [414] S. Arana-Peña, D. Carballares, R. Morellon-Sterling, Á. Berenguer-Murcia, A. R. Alcántara, R.C. Rodrigues, R. Fernandez-Lafuente, Enzyme co-immobilization: always the biocatalyst designers' choice...or not? *Biotechnol. Adv.* 51 (2021), 107584.
- [415] Mandep, P. Shukla, Microbial nanotechnology for bioremediation of industrial wastewater, *Front. Microbiol.* 11 (2020) 590631.
- [416] J. Guo, X. Liu, X. Zhang, J. Wu, C. Chai, D. Ma, Q. Chen, D. Xiang, W. Ge, Immobilized lignin peroxidase on Fe<sub>3</sub>O<sub>4</sub>@SiO<sub>2</sub>@polydopamine nanoparticles for degradation of organic pollutants, *Int. J. Biol. Macromol.* 138 (2019) 433–440.
- [417] R. Singh, U.U. Shedbalkar, S.B. Nadhe, S.A. Wadhvani, B.A. Chopade, Lignin peroxidase mediated silver nanoparticle synthesis in *Acinetobacter* sp, *AMB Express* 7 (1) (2017) 226.
- [418] X. Wen, C. Du, J. Wan, G. Zeng, D. Huang, L. Yin, R. Deng, S. Tan, J. Zhang, Immobilizing laccase on kaolinite and its application in treatment of malachite green effluent with the coexistence of Cd (II), *Chemosphere* 217 (2019) 843–850.
- [419] S. Slagman, H. Zuilhof, M.C.R. Franssen, Laccase-mediated grafting on biopolymers and synthetic polymers: a critical review, *Chembiochem* 19 (4) (2018) 288–311.
- [420] K. Horie, M. Barón, R.B. Fox, J. He, M. Hess, J. Kahovec, T. Kitayama, P. Kubisa, E. Maréchal, W. Mormann, Definitions of terms relating to reactions of polymers and to functional polymeric materials (IUPAC Recommendations 2003), *Pure Appl. Chem.* 76 (4) (2004) 889–906.
- [421] C. Yu, F. Wang, S. Fu, H. Liu, Q. Meng, Laccase-assisted grafting of acrylic acid onto lignin for its recovery from wastewater, *J. Polym. Environ.* 25 (4) (2017) 1072–1079.
- [422] V. Hahn, A. Mikolasch, J. Weitemeyer, S. Petters, T. Davids, M. Lalk, J.-W. Lackmann, F. Schauer, Ring-closure mechanisms mediated by laccase to synthesize phenothiazines, phenoxazines, and phenazines, *ACS Omega* 5 (24) (2020) 14324–14339.
- [423] D. Durán-Aranguren, J.S. Chiriví-Salomón, L. Anaya, D. Durán-Sequeda, L.J. Cruz, J.D. Serrano, L. Sarmiento, S. Restrepo, T. Sanjuan, R. Sierra, Effect of bioactive compounds extracted from *Cordyceps nidus* ANDES-F1080 on laccase activity of *Pleurotus ostreatus* ANDES-F515, *Biotechnol. Rep.* 26 (2020), e00466.
- [424] J. Su, J. Fu, Q. Wang, C. Silva, A. Cavaco-Paulo, Laccase: a green catalyst for the biosynthesis of poly-phenols, *Crit. Rev. Biotechnol.* 38 (2) (2018) 294–307.
- [425] N. Li, J. Su, H. Wang, A. Cavaco-Paulo, Production of antimicrobial powders of guaiaicol oligomers by a laccase-catalyzed synthesis reaction, *Process Biochem.* 111 (2021) 213–220.
- [426] M. Mogharabi, M.A. Faramarzi, Laccase and laccase-mediated systems in the synthesis of organic compounds, *Adv. Synth. Catal.* 356 (5) (2014) 897–927.
- [427] T. Kudanga, B. Nemađziva, M. Le Roes-Hill, Laccase catalysis for the synthesis of bioactive compounds, *Appl. Microbiol. Biotechnol.* 101 (1) (2017) 13–33.
- [428] T. Kudanga, B. Nemađziva, M. Le Roes-Hill, Laccase catalysis for the synthesis of bioactive compounds, *Appl. Microbiol. Biotechnol.* 101 (1) (2017) 13–33.
- [429] L. Pulvirenti, V. Muccilli, N. Cardullo, C. Spatafora, C. Tringali, Chemoenzymatic synthesis and  $\alpha$ -glucosidase inhibitory activity of dimeric neolignans inspired by magnolol, *J. Nat. Prod.* 80 (5) (2017) 1648–1657.

- [430] S. Dhagat, S.E. Jujjavarapu, Utility of lignin-modifying enzymes: a green technology for organic compound mycodegradation, *J. Chem. Technol. Biotechnol.* 97 (2) (2022) 343–358.
- [431] A.K. Singh, M. Bilal, D. Barceló, H.M.N. Iqbal, A predictive toolset for the identification of degradation pattern and toxic hazard estimation of multimeric hazardous compounds persists in water bodies, *Sci. Total Environ.* 824 (2022), 153979.
- [432] M. Chen, G. Zeng, Z. Tan, M. Jiang, H. Li, L. Liu, Y. Zhu, Z. Yu, Z. Wei, Y. Liu, G. Xie, Understanding lignin-degrading reactions of ligninolytic enzymes: binding affinity and interactional profile, *PLoS One* 6 (9) (2011), e25647.
- [433] N. Fu, J. Li, M. Wang, L. Ren, Y. Luo, Genes identification, molecular docking and dynamics simulation analysis of laccases from *Amylostereum areolatum* provides molecular basis of laccase bound to lignin, *Int. J. Mol. Sci.* 21 (22) (2020).
- [434] N. Fu, J. Li, M. Wang, L. Ren, Y. Luo, Genes identification, molecular docking and dynamics simulation analysis of laccases from *Amylostereum areolatum* provides molecular basis of laccase bound to lignin, *Int. J. Mol. Sci.* 21 (22) (2020) 8845.
- [435] M. Awasthi, N. Jaiswal, S. Singh, V.P. Pandey, U.N. Dwivedi, Molecular docking and dynamics simulation analyses unraveling the differential enzymatic catalysis by plant and fungal laccases with respect to lignin biosynthesis and degradation, *J. Biomol. Struct. Dyn.* 33 (9) (2015) 1835–1849.
- [436] M. Chen, G. Zeng, C. Lai, J. Li, P. Xu, H. Wu, Molecular basis of laccase bound to lignin: insight from comparative studies on the interaction of *Trametes versicolor* laccase with various lignin model compounds, *RSC Adv.* 5 (65) (2015) 52307–52313.