REACTION KINETICS OF VERSATILE PEROXIDASE FOR THE DEGRADATION OF LIGNIN COMPOUNDS

Busse, N., D. Wagner, M. Kraume and P. Czermak

Institute of Bioprocess Engineering and Pharmaceutical Technology, University of Applied Sciences Mittelhessen, Giessen, Germany
Department of Chemical and Process Engineering, Technische Universität Berlin, Berlin, Germany
Department of Chemical Engineering, Kansas State University, Manhattan KS, USA
Faculty of Biology and Chemistry, Justus-Liebig-University of Giessen, Giessen, Germany

ABSTRACT

The $\text{H}_2\text{O}_2$-dependent degradation of adlerol by a crude versatile peroxidase from Bjerkandera adusta, a new ligninolytic enzyme, was investigated. Adlerol (1-(3,4-dimethoxyphenyl)-2-(2-methoxyphenoxy)-1,3-propanediol) is a non-phenolic $\beta$-O-4 dimer whose structural architecture represents the most abundant unit (50-65%) of the valuable renewable biopolymer lignin. Lignin removal plays a key role in utilizing lignocellulosic biomass in biorefineries. Steady-state analyses in the µL scale showed saturation kinetics for both, $\text{H}_2\text{O}_2$ and adlerol with quite sensitive response to $\text{H}_2\text{O}_2$. This was characterized through slow transient states (lag phases) prior steady-state and were enhanced by increasing $\text{H}_2\text{O}_2$ concentration. The major reason for such phenomena was found to be an accumulation of compound III ($E_\text{III}$) via reaction of compound II ($E_\text{II}$) with $\text{H}_2\text{O}_2$; instead with adlerol to the enzyme’s ground state $E_\text{0}$ in order to restart another catalytic cycle. As result, the enzyme deviated from its normal catalytic cycle. A corresponding threshold was determined at $\geq 50$ µM $\text{H}_2\text{O}_2$ and an adlerol to $\text{H}_2\text{O}_2$ ratio of 15:1 for the given conditions. Furthermore, $E_\text{III}$ did not represent a catalytical dead-end intermediate as it is generally described. By an additional decrease of the adlerol to $\text{H}_2\text{O}_2$ ratio of ca. 3 at the latest, considerable irreversible enzyme deactivations occurred promoted through reaction of $E_\text{III}$ with $\text{H}_2\text{O}_2$. At a mL scale deactivation kinetics by $\text{H}_2\text{O}_2$ were further examined in dependence on adlerol presence. The course followed a time-dependent irreversible deactivation (two step mechanism) and was diminished in the presence of adlerol. The deactivation could be sufficiently described by an equation similar to the Michaelis-Menten type, competitive inhibited by adlerol. Finally, first estimates of the kinetic parameters v$_{\text{max}}$, K$_m$$_{S1}$ ($S_1$: $\text{H}_2\text{O}_2$), K$_m$$_{S2}$ ($S_2$: adlerol), k$_{\text{app}}$ and K$_{\text{app}}$ were made. Moreover, the peroxidase reaction mechanism was reviewed and recommendations are given preventing premature enzyme losses.

Keywords: Versatile Peroxidase (VP), Lignin Model Compound (LMC), Steady-State Kinetics, Slow Transient States, Deactivation

1. INTRODUCTION

Lignin is the only naturally synthesized aromatic biopolymer (Dashtban et al., 2010). Together with the polysaccharides cellulose and hemicellulose, lignin forms a complex lignin-carbohydrate network, well-known as lignocellulose, the major compound (around 50%) (Sánchez, 2009) of vascular plants (Wong, 2009), i.e. wood or straw. Lignocellulose is the most abundant renewable organic raw material on earth, and based on its main constituents of high value, with an annually production of many billion tons (Villas-Bôas et al., 2002).

Lignocellulosic material is of great interest as feedstock for bio-based industrial products and

Corresponding Author: Czermak, P., Institute of Bioprocess Engineering and Pharmaceutical Technology, University of Applied Sciences Mittelhessen, Wiesenstrasse 14, 35390 Giessen, Germany
biorefineries. Especially wood is focused on biofuel production of second generation due to several reasons: it is inexpensive, available in large amounts (Stöcker, 2008) (3,300 m³ felled or otherwise removed roundwood per year worldwide (Ek et al., 2009)) and CO₂ neutral, thus contributing to the reduction of greenhouse gas emissions (Lange, 2007). Furthermore, wood is rich in both carbohydrates cellulose (40-50%) and hemicellulose (24-35%) and in lignin ranging from 18% up to 35% (Howard et al., 2003) offering a variety of products besides the biofuels. Nevertheless, much research and development is still needed for utilizing lignocellulosic biomass (Kamm et al., 2008), such as wood, efficiently. The lignin removal plays a considerable role in this connection, since this step has to be addressed before polysaccharide bioconversion can be tackled. This process is hampered due to the lignin complexity (polydisperse 3D construct (Martínez et al., 2005) causing difficulties in analysis) (Eriksson et al., 1990) and as generally known, its enormous recalcitrance to degradation.

H₂O₂-dependent ligninolytic heme peroxidases (POXs) and O₂-dependent laccases (Lac, EC 1.10.3.2), extracellular enzymes from Basidiomycetous white-rot fungi (thus, Class II peroxidases), are the most efficient lignin degraders in nature (Kirk and Farrell, 1987). Compared to Lac, POXs have high redox-potentials (a good overview is given by Torres and Ayala (2010)) receiving “high interest as industrial biocatalysts” (Martínez, 2007), consequently. However, POXs have not yet been implemented at large scale (Torres and Ayala, 2010).

The POXs include lignin peroxidases or ligninases (LiP, EC 1.11.1.14), manganese peroxidases (MnP, EC 1.11.1.13), and versatile peroxidases (VP, EC 1.11.1.16), also known as hybrid peroxidases or lignin-manganese peroxidases, since those enzymes combine LiP and MnP catalytic properties. While LiP is probably the most famous biocatalyst studying delignification processes, VP is a relative new POX and was first interpreted as a MnP in 1996 by Martínez et al. and in 2000 by Giardina et al. (Ruiz-Dueñas et al., 2009a). In contrast to LiP and MnP, VPs are capable to direct degradation/oxidation of a broad spectrum of persistent substrates (e.g. (non-) phenolic lignin compounds, dyes, such as RB5 and others) meaning without the requirement of mediators (Pogni et al., 2005); an important feature for biotechnological applications. Although VPs are limited available (a major drawback for industrial utilization), they have still attracted double attention, “as model enzymes and as a source of industrial/environmental biocatalysts”, within the last years (Ruiz-Dueñas et al., 2009a). Nonetheless, research work for delignification concerning VPs are seldom, in particular kinetic studies with non-phenolic lignin model dimers (lignin model compounds (LMC)) containing a β-O-4 linkage. These components are the most frequent structural units in lignin biopolymers ranging from 50% up to 65% (Adler, 1977). Moreover, the cleavage of such LMC is a substantial indicator for enzyme catalyzed lignin depolymerization processes (Tien, 1987; Wong, 2009), β-O-4 dimers are therefore suitable for studying delignification mechanisms in a more or less simplified manner as it will be seen later in the text.

The present work focuses on kinetic investigations based on the degradation of the β-O-4 lignin model dimer adlerol by a new (Jena Bioscience, 2010) crude versatile peroxidase (VP) from Bjerkandera adusta as a ligninolytic model peroxidase. All studies were conducted without the need of any mediator (e.g. the non-phenolic monomer veratryl alcohol (VA) or Mn²⁺) or surfactant like Tween. In addition, the pH optimum of reaction was determined and enzyme deactivation by H₂O₂ depending on adlerol amount was also examined.

Ligninolytic peroxidase assays are generally known to be more complex and difficult to optimize (Sinsabaugh, 2010; German et al., 2011). Prior starting with the experimental description, the paper will therefore be continued by a compact review of the POX reaction mechanism providing basic information for a better understanding and in order to differentiate pure enzymatic from non-enzymatic reactions. For this purpose most background knowledge was received from authors examining horseradish peroxidase (HRP) activity from plants (Class III peroxidases) on appropriate phenolic substrates as well as LiP using mainly VA as substrate; β-O-4 lignin model dimers are also involved as LiP substrates. VA is a second metabolite of Phanerochaete chrysosporium and other white-rot fungi (Schoemaker and Piontek, 1996) and thus the mediator of choice for LiP enabling phenolic compound oxidation. Both, LiP and HRP are similar in many characteristics (e.g. the enzyme oxidation states as depicted in Fig. 1-3) (Schoemaker, 1990) and are so far the most popular and best studied peroxidases. Moreover, it will be expected that the used VP should be in compliance with a LiP from P. chrysosporium at least for ca. 60%
(Ruiz-Dueñas et al., 1999). Consequently, LiP reactions are more concerned for comparison throughout this study than those for MnP. Additionally, MnP is strongly Mn²⁺-dependent resulting in Mn-mediated degradation processes.

2. THE REACTION MECHANISM

It is widely adopted that the catalytic peroxidase (POX) cycle follows a ping-pong mechanism. In contrast to the classical mechanism, the POX reaction scheme (“common for most heme peroxidases” (Dunford, 2010)) is assumed to be irreversible as sketched in Fig. 1 in simplified terms. That means, formation of enzyme substrate complexes (e.g. ES) prior reaction product generation can be neglected at this point, since they are assumed to be of fleeting non-detectable existence (Dunford, 1991).

Within its catalytic cycle in Fig. 1 the enzyme undergoes two fundamental structural changes. First (pathway 1), the reaction will be initiated by hydrogen peroxide (H₂O₂) through a pH-independent (within the range of approximately 2-7.5 for a LiP (Andrawis et al., 1988) or 3.5 and 7 for a VP (Pérez-Boada et al., 2005)) two-electron oxidation converting the enzyme from its native resting ferric/ground state (E₀) to the so-called compound I (E₁, first intermediate) (Equation (1)), a protein cation radical (Torres and Ayala, 2010) of strong oxidative power (Arnao et al., 1990b).

\[
E^0 + H_2O_2 \underset{k_1}{\rightarrow} E^1 + H_2O \tag{1}
\]

k: reaction rate constant; index: pathway number

Subsequently, E¹ will be reduced back to the resting state E₀ via a second enzyme intermediate (E₁I, compound II) through two consecutive one-electron reduction steps triggered by suitable reducing substrates (S, the actual electron donor “preferentially electron-rich aromatic compounds” (Lundell et al., 1993a)). Both reaction steps are pH-dependent (Pérez-Boada et al., 2005; Wong, 2009) and causing release of radical cation intermediates (S⁺⁺) (Palmer et al., 1987) (pathway 3 and 4 in Fig. 1, and Equation (2)-(3)):

\[
E^1 + S \underset{k_3}{\rightarrow} E^{II} + S^{**} \tag{2}
\]
\[
E^{II} + S \underset{k_4}{\rightarrow} E^0 + S^{**} + H_2O \tag{3}
\]

with \( k_3 >> k_4 \).

Substrates causing E¹ reduction in Equation (2) can also reduce E₁I (Equation (3)) as described by Dunford (2010) for HRP. The same applies for ligninolytic enzymes, such as LiP, and phenolic substrates (Schoemaker, 1990). Hence, the overall net enzyme reaction can be expressed according to Equation (4).

\[
2S + H_2O_2 \quad \rightarrow \quad 2S^{**} + 2H_2O \tag{4}
\]

The consumption rate \( r \) for S and H₂O₂ can be defined as follows.

\[
r = -\frac{d[S]}{dt} = -\frac{1}{2} \frac{d[H_2O_2]}{dt} = \frac{d[S^{**}]}{dt} \tag{5}
\]

However, caution should be exercised with regard to generalization once non-phenolic substrates are involved.

Compound II (E₁I) of LiP can just be reduced by dimethoxylated (minimum amount of alkoxy substituents) non-phenolic aromatic compounds, whereas E¹ also accepts mono-methoxylated aromatic components (Schoemaker, 1990).

Fig. 1. Simplified “POX ping-pong” mechanism. E₀-E₁I represents the three main peroxidase oxidation states. The symbols S and S⁺⁺ stand for an appropriate substrate (e.g. adlerol) and its corresponding radical cation, respectively. The reaction will be initiated by H₂O₂ (pathway 1) followed by two consecutive one-electron reduction steps (pathway 3-4).
The radical cation intermediates will further pass through non-enzymatic reactions (diffusion controlled) yielding complex mixtures with a wide variety of final products. Depending on the nature of S', the subsequent non-enzymatic reactions include (based on Palmer et al. (1987) and Schoemaker (1990)):

- one-electron oxidation of another appropriate substrate molecule (e.g. lignin or lignin derivatives) while S' will be reduced to its ground state (S) (Palmer et al., 1987); this reaction is also referred to as mediator (diffusible oxidizer) behavior and may be stimulated through electronic transfer from the reducing substrate to S' (Schoemaker, 1990)
- S' may also degrade immediately to a free radical R' (R also stands for aromatic residual) (Palmer et al., 1987) caused by rapid deprotonation processes (Schoemaker et al., 1994a)
- side-chain cleavage (e.g. Cα-Cβ bond cleavage as seen in Fig. 2, C-H bond cleavage)
- demethoxylation
- ether-bond cleavage (e.g. addition of solvent (H2O)) (Palmer et al., 1987)
- hydroxylation (e.g. through oxygen incorporation via O2 or H2O from the solvent) of benzylic methylene groups
- phenol formation (by nucleophilic attack, e.g., addition of solvent (H2O) (Palmer et al., 1987))
- aromatic ring cleavage caused by reactions with perhydroxy radicals (HO2'; details follow in the next paragraph) (Palmer et al., 1987)

The free-radical species (R') are highly reactive (Palmer et al., 1987) which in turn can bind molecular oxygen (O2) forming the final product and a superoxide anion radical (O2−) (Schoemaker, 1990) via degradation of an organic peroxy radical intermediate (ROO·) (Palmer et al., 1987). O2− undergoes rapid disproportionation to H2O2 and O2 (Harman et al., 1986; Schoemaker, 1990) (Fig. 3, Equation (b)). The O2− is in a pH-dependent equilibrium with its protonated counterpart, the perhydroxy radical (HO2·) (Bielski et al., 1985; Fig. 3, Equation (a)). At low pH conditions HO2· predominates and represents a powerful oxidant (Halliwell and Gutteridge, 1985), thus contributing to substrate degradation processes by initiating additional bond cleavage in consequence of substrate (S) oxidation to its radical cation. The oxidant is itself reduced to H2O2 (Palmer et al., 1987).

ROO· can react with a further substrate molecule, alternatively, resulting in a new free radical and an organic peroxy (ROOR·; if R’ stands for hydrogen, organic hydroperoxide (ROOH) will be produced) formation (Palmer et al., 1987). Moreover, R’ can undergo dimerization (reaction with other free radicals) as well as polymerization processes (reaction with neutral molecules) (Palmer et al., 1987). In particular, phenolic structures “are prone to polymerize under oxidative conditions” (Schoemaker, 1990).

Free-radical products may also attack the peroxidase along with an inactivation process (Nicell et al., 1993) on the one hand or they act as an additional suitable substrate molecule (thus the radicals can be also considered as S (depending on their chemistry)) for maintaining the catalytic cycle on the other hand. The latter is partly applicable for the radical cation intermediates as discussed later in the text.

Next, the basic adlerol degradation steps, induced by (fungal) heme peroxidases, will be explained aiming to meet the needs of the current study. The graphical illustration is given in Fig. 2. Here, the catalytic cycle will be initiated again by H2O2 to form E1·. E1 further reacts with adlerol (structure I) to E1· and an adlerol cation radical intermediate (structure II) (Lundell et al., 1993b) which will be rapidly fragmented non-enzymatically between its Cα-Cβ bonds in the subsequent (Hatakka et al., 1991). For the latter, various theories have been under consideration.

Based on Tien and Kirk (1984), Kirk et al. (1986) and Hatakka et al. (1991), the consequence by LiP action is the Cα-Cβ cleavage resulting mainly in veratraldehyde (VALd) (structure IV) formation from the Cα moiety. In parallel, formation of a Cβ-centred radical (structure VIII) was presumed leading to guaiacol (structure VII) and glycoaldehyde (structure VI) through reaction with O2 and eventually via an unstable hemiacetal intermediate (structure IX) (Kirk et al., 1986). In general, the reaction course was suggested to be similar to β-1 lignin model compounds (Tien and Kirk, 1984). In contrast, Lundell et al. (1993b) and Schoemaker et al. (1994b) concluded a slightly different non-enzymatic reaction after Cα-Cβ cleavage assuming structure II (in Fig. 2) with a cation radical centre at the Cβ-ether oxygen position as consequence of LiP action. They were able to show formation of a veratryl alcohol radical (VA·, structure III) and a Cβ cation (structure V) reacting to component IV (preferably), VI and VII, respectively.
Fig. 2. Graphical illustration of the adlerol (β-O-4 model compound, structure I) biocatalysis based on LiP. Several degradation variants (partly in a summarized manner) are shown derived from proposed reaction schemes by Tien and Kirk (1984); Kirk et al. (1986); Lundell et al. (1993b) and Schoemaker et al. (1994b). A detailed description is found in the text.
In addition to the Ca-Cβ cleavage, the adlerol cation radical can also undergo Ca-oxidation (e.g. due to proton loss (Kirk et al., 1986; Schoemaker, 1990) or direct hydrogen abstraction in the presence of active oxygen species (Snook and Hamilton, 1974) giving rise to a corresponding ketone (structure X, second major product (15%)’ generation) (Tien and Kirk, 1984; Kirk et al., 1986). For this purpose, reaction schemes are published by Schoemaker (1990) and by Kirk et al. (1986).

Furthermore, some more compounds (e.g. veratrylglycerol) could be detected by Kirk et al. (1986), but just in small amounts (thus, these components may be omitted in further analyses). The product distribution of the β-O-4 lignin model compound. This may be an important feature because lignin has a highly irregular structure.

Under substrate (adlerol) limited conditions Tien and Kirk (1984) demonstrated a non-stereospecificity of their used ligninolytic enzyme (LiP) due to complete cleavage of the β-O-4 lignin model compound. This may be an important feature because lignin has a highly irregular (Schoemaker, 1990) structure.

As already indicated above, Fig. 1 and 2 give just a first insight into the topic. Indeed, the mechanism is far more complex considering certain circumstances (e.g. reaction conditions, enzyme type, presence of inhibitors, substrate and radical chemistry, respectively) as shown schematically in Fig. 3. Hence, in the interest of simplification, all POX intermediates are pictured in a more general representation, as already done by several authors, for this study (due to some structural differences between HRP and ligninolytic enzymes, like VP). In case of the used VP, EI and EI1 are expected to be tryptophan radical containing proteins. Thereby, the said tryptophan radical is suggested to be located at the enzymes surface (~ 10-11 Å to the heme centre (Pogni et al., 2005; Ruiz-Dueñas et al., 2009a)) for substrate oxidation purposes. That means, there is no direct contact between the heme (FeII) co-factor and the reducing aromatic substrates, ranging from VA (non-phenolic monomer) up to the complex lignin biopolymer (macromolecule), avoiding steric hindrance. Interactions between substrate and the heme group will be enabled via long range electron transfer (LRET) (Ruiz-Dueñas et al., 2009a). The involvement of LRET in lignin degradation processes through ligninolytic enzymes seems to be generally accepted and can be read up elsewhere.

In the following, a brief explanation will be given for each reaction pathway in Fig. 3.

Based on investigation results of transient state kinetics using a recombinant native VP (obtained by E. coli), both EI and EI1 reduction by VA oxidation exhibited reactions which can be described by Equation (6) and (7) (Ruiz-Dueñas et al., 2009b):

\[ \text{E}^1 + S \leftrightarrow K_{D1} \rightarrow \text{E}^1 - S \xrightarrow{k_3} \text{E}^{11} + S^* \]  
(6)

\[ \text{E}^{11} + S \leftrightarrow K_{D4} \rightarrow \text{E}^{11} - S \xrightarrow{k_4} \text{E}^0 + S^* \]  
(7)

KDi and ki (with i = 3, 4) are the corresponding equilibrium dissociation constant and rate constant, respectively. That implies, reversible steps (formation of an enzyme-substrate complex or also defined as precursor complex (Dunford, 2010)) are certainly possible within pathway 3 and 4 in Fig. 1-3. A reaction behavior similar to Equation (7) was also proposed for the ABTS ([2,2’-azino-bis[3-ethylbenzthiazoline-6-sulfonic acid] diammonium salt] oxidation by a HRP (Smith et al., 1992). As a result, Smith et al. (1992) concluded the product dissociation as limiting step, most likely due to good electron donor properties of ABTS which are beneficial for fast precursor complex development.

The two single one-electron oxidations of the substrate S described through Equation (2) and (3) are not universal for peroxidases (Kanofsky, 1991). In case of certain substrates, the two-electron oxidation shown in pathway 9 of Fig. 3 (van Rantwijk and Sheldon, 2000)
could be dominant as demonstrated with a thyroid peroxidase (TPO) by Nakamura et al. (1985).

Furthermore, it seems possible that both, one- and two-electron transfer routes, may occur within a catalytic cycle (Nakamura et al., 1985). For ligninolytic systems, this phenomenon cannot be excluded for sure (this point is also discussed by Schoemaker (1990)). Additionally, an alternative reduction of EIII by radicals (component III), as depicted in Fig. 2, makes it difficult to differentiate between two serial one-electron steps and one single two-electron step (Schoemaker, 1990).

However, the most important (side) reactions in the catalytic cycle appear once the S to H2O2 ratio is unbalanced (Arnao et al., 1990a; 1990b). It is well-known that the co-substrate H2O2 is essential for the catalytic cycle but in excess it will favor reactions diminishing enzyme activity (pathway 5 in Fig. 3 (Arnao et al., 1990b; Chung and Aust, 1995)) up to enzyme inactivation (Ei) (pathway 10, 11 and 13 (Arnao et al., 1990b), and pathway 14 (Wariishi and Gold, 1990)). At low S/H2O2 ratios both substrates compete for reactions with Ei (Tien, 1987; Arnao et al., 1990a) and EIII (Chung and Aust, 1995).

In the event of large H2O2 excess or in the absence of a suitable reducing substrate, the POX exhibits time-dependent irreversible inactivation (Vlasits et al., 2010) kinetics (combination of pathway 10, 11 and 13 in Fig. 3). Such inactivation is designated as suicide inhibition in the literature belonging to mechanism-based inhibitions. For detailed information the reader is referred to Copeland (2002) and Marangoni (2003). Thereby, the intermediate E′−H2O2 (may also be of fleeting existence (Dunford, 1991)) must not lead exclusively to an inactivated enzyme Ei (pathway 13).

Several pathways are possible.

Concerning the backward reactions first, E′−H2O2 is reversibly dissipated to Ei (pathway 11) enabling H2O2 consumption to H2O and O2 yielding a reduction to E0 (pathway 2 (Nakajima and Yamazaki, 1987; Valderrama et al., 2002; Vlasits et al., 2010)). Therefore, reaction path 2 is related to catalase activity (Arnao et al., 1990b; Vlasits et al., 2010), whereas Arnao et al. (1990b) referred to a direct reaction of E′−H2O2 with H2O2 (in the absence of reducing substrate) in this context. E′−H2O2 can also be transformed to EII (under superoxide anion radical (O2•−) release; pathway 12 (Arnao et al., 1990b)), which will further react with H2O2 (pathway 5) to another third intermediate EIII (also defined as compound III) of none or far less catalytic activity (Schoemaker, 1990). This EIII formation will be affected by pH (beside the S/H2O2 ratio) and is enhanced at acidic pH values (Cai and Tien, 1992). Finally, EIII becomes either irreversibly inactivated (pathway 14, mediated by further pH-dependent reactions with H2O2 (Wariishi and Gold, 1990; Goodwin et al., 1994)) or it decomposes spontaneously (Nakajima and Yamazaki, 1987) under superoxide radical release to the ferric state EIII (slowly unimolecular reaction, pathway 7 (Arnao et al., 1990b)). Vice versa, Wariishi and Gold (1990) also documented a conversion of EIII to EII by O2− in their catalytic scheme with LiP (reverse reaction of pathway 7 (Fig. 3)). Alternatively, if reducing substrate is present, EIII returns to Ei via pathway 6 (reaction is obviously faster than in pathway 7 (Cai and Tien, 1992)) generating a radical cation (S•+) (as seen in reaction pathway 3 and 4 (Fig. 1-Fig. 3)) and H2O2 (Yokota and Yamazaki, 1965; Acosta et al., 1988). Moreover, it is reported that EIII may be converted to Ei by an one-electron oxidation of S (Tamura and Yamazaki, 1972) as explained through pathway 8. At this point, it has to be mentioned that not all potential POX substrates (S as well as related radicals) are capable to revert EIII (basically to E0) necessarily. Consequently, EIII remains catalytically inactive and will be subjected to oxidative stress due to reactions with H2O2. A practical example for such phenomena is given by Chung and Aust (1995) studying the inactivation of the isozyme H2 of LiP from P. chrysosporium by H2O2 during phenol oxidation in comparison to VA. They found out that neither the phenolic substrate nor its free-radical product (phenoxyl radical) was competent to convert EIII back to Ei. The same was observed for VA contrary to its radical cation product (VA•+), which reverted EIII to native enzyme as marked by 6′ in (Fig. 3).

For EIII of HRPs an additional decomposition to EII was supposed by H2O2 dissociation (pathway 19) (Tamura and Yamazaki, 1972; Nakajima and Yamazaki, 1987) through reaction with reducing substrate (Cai and Tien, 1992).

Reconsidering the start-up from the ferric state Ei in Fig. 3, an alternative way was introduced by Acosta et al. (1988) (originally worked out by Smith et al. (1982)) via direct reaction with substrate S forming the ferrous enzyme (E′) under S•− release (pathway 15). Depending on the presence of O2, two subsequent reaction variants of E′ may occur. On the one hand E′ is oxidized to EIII (pathway 16) independent of the O2 level (Smith et al., 1982; Acosta et al., 1988), on the other hand E′ will be converted to EII by H2O2 in the absence of O2, releasing H2O (pathway 18) (Jantschko et al., 2005; Dunford, 2010).
Fig. 3. Summary of possible peroxidase intermediates, their development and reaction pathways collected from various research reports containing enzyme reaction studies with horseradish peroxidases and ligninolytic peroxidases (mostly LiP) as well for different reducing substrates. The normal peroxidase cycle is highlighted through bold solid lines while its involved enzyme intermediates are color-shaded. The reaction pathways are numbered (1-19) for identification purposes. The representation of each enzyme intermediate/compound was derived from several authors (Arnao et al., 1990; Schoemaker et al., 1994; Dunford, 2010; Torres and Ayala, 2010) showing formal changes at the heme group. Equation (a)-(e) introduce several important non-enzymatic reactions for completeness, since organic hydroperoxide ROOH competes for reaction with $E_0^0$ and $E^0$. As a result, enzyme inactivation (pathway 13) occurs (Acosta et al., 1988).

\[
\begin{align*}
\text{HO}_2^+ & \rightleftharpoons H^+ + O_2^- & (a) \\
2 H^+ + 2 O_2^- & \rightarrow H_2O_2 + O_2 & (b) \\
S^{2-} & \rightarrow H^+ \rightarrow R' & (c) \\
R' + O_2 & \rightarrow R'OO' + CO_2 & (d) \\
R'OO' + RH & \rightarrow R'OOH + R' & (e)
\end{align*}
\]
Thus, Acosta et al. (1988) gave another option for initiating the catalytic cycle without H$_2$O$_2$ addition. Smith et al. (1982) reported also a return of E$_{III}$ to E$^\prime$ described through pathway 17 (Acosta et al., 1988).

To gain a deeper insight into the structural changes of the enzyme due to reactions shown in Fig. 3, the reader is referred to Dunford (1991; 2010) as well as to Torres and Ayala (2010).

Finally, superoxide (or perhydroxyl radicals HO$_2^\cdot$ at low pH) can be additionally served as electron donor, instead of a reducing substrate. Consequently, all three enzyme states E$^I$, E$^{II}$, and E$^{III}$ may be reduced by O$_2^\cdot$ (such unimolecular reactions are omitted in Fig. 3 for better clarity) under O$_2$ production (Bielski et al., 1985; Kettle et al., 2007) according to Equation (8)-(11), based on Kettle et al. (2007).

$$E^I + O_2^- \rightarrow E^{II} + O_2 \quad (8)$$

$$E^{II} + O_2^- + H^+ \rightarrow E^0 + O_2 + H_2O \quad (9)$$

$$E^{III} + O_2^- + 2 H^+ \rightarrow E^0 + H_2O_2 + O_2 \quad (10)$$

or

$$E^{III} + O_2^- + 2 H^+ \rightarrow E^I + H_2O + O_2 \quad (11)$$

The reaction in Equation (9) is supposed to prevent enzyme inhibition caused by poor reducing substrates for maintaining its activity (Kettle et al., 2007). In contrast, superoxide and reducing substrate S compete for reactions with the enzyme. Similarly, competition between H$_2$O$_2$ and organic hydroperoxides of the form ROOH (Fig. 3, Equation (e)) also exists for reaction with E$^{II}$ to E$^I$ and between S and ROOH for E$^I$ conversion depending on the relation of their concentrations (reaction pathways are also omitted in Fig. 3 for more clarity). Reactions of ROOH with E$^I$ lead to enzyme inactivation. At low O$_2$ concentrations ROOH formation is limited (Acosta et al., 1988).

3. MATERIALS AND METHODS

In the following all represented concentration data are final concentrations.

3.1. The Enzyme and its Preparation

The heme peroxidase used in this study was a lyophilized crude versatile peroxidase (VP) from B. adusta, purchased from Jena Bioscience GmbH, Germany, with a molecular weight (MW) of 43 kDa. The lyophilisate was generally stored at -20°C. For each experiment a fresh VP stock solution, with distilled water as solvent, was prepared directly from the lyophilisate. The Reinheitszahl (RZ) value (ratio heme to total protein content) was sporadically checked by recording the absorbance of 200 µL stock solution at 407 nm (maximum absorption of the heme group or soret band for that VP) and 280 nm (total protein) at room temperature (RT). For this, a HELLMA quartz glass microplate (supplied by VWR International GmbH, Germany) and a BioTek microplate reader Synergy HT (BioTek, Germany) was used. The RZ value for the crude VP was 0.3. At least a factor of ten lower, than RZ values for other purified VPs, ranging from 3.2 (Moreira et al., 2006) and 3.5 (Pogni et al., 2005) up to 4.0 (Garcia-Ruiz et al., 2012). However, the RZ value is no indicator for enzymatic activity necessarily (Dunford, 1991; SAC, 2012).

Enzyme activity measurements were carried out according to the instructions, as seen below. Depending on their outcome a further dilution of the enzyme sample to an adequate working solution was required.

With regard to ensuring that enzyme losses could be excluded and the same starting point could be warranted for all studies, respectively, regular enzyme activity measurements of the used VP solution were performed at the beginning as well as at the end of each experimental day. All VP solutions showed persistent activity (1 day) by storing in distilled water at RT.
3.2. Preparation of the Co-Substrate Hydrogen Peroxide (H$_2$O$_2$)

Hydrogen peroxide (30% EMSURE® for analysis from Merck KGaA) was purchased from VWR International GmbH, Germany. For each experimental day a suitable stock solution was prepared freshly by diluting the 30% solution with distilled water. To prevent H$_2$O$_2$ consumption, i.e. caused by diverse contaminations or light irradiation, prior enzyme reactions start, the reservoirs needed were cleaned and flushed thoroughly with distilled water before use. Furthermore, the reservoirs were kept sealed, if possible, and the stock solutions were stored in the dark, additionally.

The concentration of each stock solution was controlled by measuring the absorbance at 230 nm ($\epsilon_{230nm} = 0.078$ mM$^{-1}$ cm$^{-1}$) in a HELLMA 104-QS semi-micro cuvette SUPRASIL® 10.00 mm (obtained from VWR International GmbH, Germany) using a Thermo Spectronic HELIOS Y UV-Visible spectrophotometer (Thermo Fisher Scientific, Germany). At random intervals repeated concentration measurements were done for H$_2$O$_2$ stability evaluation. All rechecked solutions were stable.

Depending on the experimental design a further dilution of the stock solution with distilled water to a suitable working solution were also necessary.

3.3. Enzyme Activity Measurements

3.3.1 The Assay Procedure

For determining the VP activity, the well-known oxidation reaction of ABTS ([2,2'-azino-bis(3-ethylbenzthiazoline-6-sulfonic acid)] diamonium salt) (purchased from AppliChem GmbH, Germany) to the stable green cation-radical ABTS$^•+$ (Barr and Aust, 1994) was monitored at $\lambda = 420$ nm according to an internal standard procedure. The assay was carried out in NUNC Immuno-Modules, F8 Polysorp (supplied by VWR International GmbH, Germany) and again on the BioTek microplate reader Synergy HT for the photometric measurements. The reader was operated and programmed through the corresponding data collection and analysis software Gen5™, version 1.06.

Each activity test was conducted in sodium acetate buffer at 25°C. The final reaction volume was 200 µL resulting in a layer thickness of 0.57 (determined according to a technical note of Promega (2009)). VP activities were expressed in units (U). Thus, 1 U was defined as the enzyme amount forming 1 µmol product (ABTS$^•+$) from ABTS per minute. The enzyme activity (U mL$^{-1}$) was calculated using the Gen5 software and its well analysis calculation type Max V, value of the maximum slope (maximal change in absorbance ($\Delta$E) in OD min$^{-1}$), in Lambert-Beer’s law. Every maximum slope was generated from a data set of 8 measuring points along a straight line of $R^2 \geq 0.998$.

Each enzyme activity measurement was performed in triplicates. The relative error of the assay was 2-7%.

3.3.2. Preparation of the Sodium Acetate Buffer

The used buffer system was composed of 1 M acetic acid (obtained from 100% acetic acid, EMSURE® anhydrous for analysis from Merck KGaA and purchased by VWR International GmbH, Germany) and 1 M sodium hydroxide (made of sodium hydroxide pellets purchased from Carl ROTH GmbH and Co. KG, Germany). The pH was adjusted by titrating with 1 M NaOH while stirring at RT. Finally, the buffer was sterile-filtered with a 0.22 µm PES filter (250 mL polyester sulfone filter from Biochrom AG, Germany) and stored at 2-8°C in the refrigerator until use. For each experimental day the buffer was incubated over night at RT.

3.3.3. Preparation of an ABTS Working Solution

A working solution, varying from 20 to 50 mL in volume according to requirements, was obtained by solving ABTS in distilled water. Finally, it was kept refrigerated (2-8°C) in 1 mL aliquots (1.5 mL Rotilabo® reaction vessels for light-sensitive samples from Carl ROTH GmbH and Co. KG, Germany) until use. Under these conditions the working solution is stable at least for one month based on experience. Before each examination the amount of aliquots needed were brought to RT.

3.4. The Adlerol Assay for Studying VP Kinetics

3.4.1. The Experimental Procedure

The kinetic assay (modified based on Tien and Kirk (1984)) was carried out in a HELLM A quartz grass microplate and on a BioTek microplate reader Synergy HT for monitoring the oxidation product veratraldehyde (V Ald) (3,4-dimethoxybenzaldehyde) at $\lambda = 310$ nm ($\epsilon_{310} = 9300$ M$^{-1}$ cm$^{-1}$ (Tien and Kirk, 1984)) at 30±1°C. The reaction mixtures were generally composed of approximately 0.3 mg mL$^{-1}$ lyophilisate, 0.1 M sodium tartrate buffer (pH 4.0), H$_2$O$_2$ and adlerol varying from
0.015 mM to 0.15 mM and 0.15 mM to 1.5 mM, respectively. For this purpose, the enzyme (0.3 mg mL\(^{-1}\) final concentration) was pre-incubated first in the microplate reader, together with adlerol and buffer (thus, in an incomplete mixture), at an adjusted temperature of 35°C. The reaction was initiated by H\(_2\)O\(_2\) addition once a temperature of 30°C could be measured in the test solution (external temperature measurement device equipped with a Pt100 sensor from Testo AG, Germany). As a result, 7 min were necessary in principle for pre-incubation.

Preliminary studies have shown that the temperature control unit of the microplate reader has to be adjusted to 35°C in order to maintain a desired temperature of 30±1°C in the reaction mixture with a total volume of 200 µL (resulting in a layer thickness of 0.57 which was determined according to a technical note of Promega (2009)). The actual reaction time and the time interval for recording UV absorption of VAld were 4 min and 10 sec. An enzyme-adlerol mixture (final adlerol concentration varied as seen above) was used as blank.

Controls have taken place previously to evaluate adlerol decay in absence of the VP or H\(_2\)O\(_2\), respectively. It was found that adlerol could be only degraded to VAld in the presence of both, VP and H\(_2\)O\(_2\).

All examinations were performed in triplicates. Hence, the reaction velocities (calculations similar to ABTS assay above) were determined as the mean of these three measurements with a maximal standard deviation of ≤ 10% from the mean.

3.4.2. Determination of the pH Optimum

The experimental set-up for determining the pH optimum was similar to the kinetic assay. The major differences in this study are the fixed adlerol and H\(_2\)O\(_2\) concentration of 1 mM and 0.1 mM, respectively. In addition, the pH of the 0.1 M sodium tartrate buffer was varied in 0.5 pH units, ranging from 2.0 to 5.0. Buffer solutions were not sterile-filtered because of the direct use after preparation.

3.4.3. Preparation of the Sodium Tartrate Buffer

The buffer system, pH 4.0, was made up of 1 M tartaric acid (obtained from L(+) tartratic acid, EMSURE\textsuperscript{®} for analysis from Merck KGaA and purchased by VWR International GmbH, Germany) and 1 M sodium hydroxide, similar to the sodium acetate buffer preparation above.

3.4.4. Preparation of an Adlerol Stock Solution

Adlerol (1-(3,4-dimethoxyphenyl)-2-(2-methoxy-phenoxy)-1,3-propanediol), with a molecular weight of 334.36 g mol\(^{-1}\), was purchased from Wako Chemicals GmbH, Germany (manufacturer is Wako Pure Chemical Industries, Ltd. in Japan). For the kinetic investigations a stock solution was made by solving the required adlerol amount with distilled water containing 10% (v/v) acetone under vigorous stirring for 2-3 h at 35°C. Subsequently, it was stored in a sealed amber glass vial, and in the dark, at RT until use. Under these conditions the adlerol solution is stable at least for one month based on experience.

Depending on examination conditions, the stock solution must be further diluted with distilled water to an appropriate working solution prior starting the experiment.

3.5. Set-up for Studying VP Deactivation by H\(_2\)O\(_2\)

Enzyme deactivation studies were conducted in a 10 mL and 50 mL scale, respectively, under continuously stirring with a speed of n = ca. 375 rpm at 30±1°C using a magnetic stirrer of the type MR Hei-Standrad, from Heidolph Instruments GmbH and Co. KG, Germany. For this purpose, an average lyophilisate concentration of 0.06 mg mL\(^{-1}\) (final concentration with a maximal standard deviation of < 10%) was incubated with H\(_2\)O\(_2\) varying from initially 0 mM up to 1 mM and in the presence (4 mM (10 mL scale)) or absence (50 mL scale) of adlerol. Reaction conditions (buffer system, pH, practical procedure) were similar to those seen in the section above. The reaction was started by enzyme addition and VP stability was determined every three minutes, through the internal ABTS assay, over a time period of 18 min. To this, the measuring time of the ABTS assay has to be modified without affecting signal linearity for reaction velocity determination.

All examinations were performed in duplicates. The remained active enzyme concentration was therefore determined as the mean of these two measurements with a maximal standard deviation of 10% from the mean.

4. RESULTS

4.1. Effect of pH on VP Reaction Rate

The pH optimum for the H\(_2\)O\(_2\)-dependent adlerol degradation to the major product veratraldehyde (VAld)
by the crude VP from B. adusta appeared within the range of 3.5 and 4.0 (Fig. 4).

4.2. Influence of the Pre-incubation Phase with Adlerol

As introduced throughout section “Materials and Methods” the used VP was pre-incubated with adlerol prior each reaction start by H$_2$O$_2$ in order to bring up the reaction temperature to 30°C. The pre-incubation phase was continuously monitored in the incomplete reaction mixture (none H$_2$O$_2$, thus the final volume was not achieved without causing any considerable impact) by recording the absorption at 310 nm every 10 sec. It was found that the absorption showed saturation behavior while the time course was clearly affected by adlerol concentration, examples are illustrated in Fig. 5. The time required reaching saturation was reduced by an adlerol increase. Furthermore, after saturation was achieved, a relative slow but constant decrease in absorption started; with some time delay at the higher adlerol concentration.

To gain a better understanding of these findings the soret band (heme group absorption maximum of the used VP at 407 nm) was also investigated independently, under equal conditions, at adlerol concentrations of 0 and ca. 1 mM (Fig. 6). Any red or blue shift in the soret band are indicators for changes in the enzyme transition state (E$^0$, E$^I$-E$^III$ as well as E$^f$). Those shifts are quasi specific and each transition state can be assigned by a certain wavelength, thus enabling the identification of conformational changes of the enzyme under study. For initial orientation, the given characteristic wavelengths for E$^I$-E$^III$ in Table 1 (received from literature) were assumed for this work. Moreover, in order to obtain a representative reference for E$^0$ and E$^III$, the soret band was also examined by storing the VP just in distilled water and 0.1 mM H$_2$O$_2$, respectively. Both, in the absence of adlerol. On this basis, (notwithstanding the fact that E$^II$ and E$^III$ may be difficult to differentiate, which will be further hampered by the chosen buffer system) it is clearly seen in Fig. 6, that the VP was initially in its oxidation state E$^{III}$ (by comparing the black dashed solid line with the red ones (VP in H$_2$O$_2$)) before it was transferred to its ground state E$^{II}$ over pre-incubation time; regardless of adlerol presence. This observation is in good agreement with Fig. 5. That means, E$^{III}$ spontaneously decayed to E$^{II}$.

![Fig. 4. Effect of the pH on the enzymatic H$_2$O$_2$-dependent adlerol conversion to veratraldehyde. The study was performed in triplicates in 100 mM sodium tartrate buffer with 100 µM H$_2$O$_2$, 1.0 mM adlerol and ca. 0.3 mg mL$^{-1}$ VP lyophilisate at 30°C.](image)

![Fig. 5. A-B: Effect of the adlerol amount during pre-incubation. The enzyme was pre-incubated in ca. 100 mM sodium tartrate buffer, pH 4.0 at 0 µM adlerol (reference) ( ), 0.13 mM adlerol ( ) and 1.3 mM adlerol ( ). The last two were conducted in 133 mM sodium tartrate buffer, pH 4.0, since the mixtures were without H$_2$O$_2$ and thus, still incomplete in volume.](image)
Table 1. Assumed soret band maxima of the intermediates $E_0$ - $E_{III}$ for orientation purposes.

| Enzyme state | $\lambda$ nm | Enzyme | Reference |
|--------------|--------------|--------|-----------|
| $E_0$        | 407          | VP$^a$ | Pogni et al. (2005) |
| $E_1$        | 403          | rVP$^b$ | Pérez-Boada et al. (2005) |
| $E_{II}$     | 417          | rVP$^b$ | Pérez-Boada et al. (2005) |
| $E_{III}$    | 420          | LiP$^c$ | Tien and Kirk (1984) |

$a$. from *Bjerkandera adusta*  
$b$. recombinant VP from *Escherichia coli*  
$c$. from *Phanerochaete chrysosporium*

Fig. 6. Behavior of the soret band within the first 6 minutes of pre-incubation. A: VP in 100 mM sodium tartrate buffer, pH 4.0, B: VP incubation in an incomplete reaction mixture (without H$_2$O$_2$) in the presence of ca. 133 mM sodium tartrate buffer, pH 4.0 and ca. 1.3 mM adlerol. The black dashed line represents the starting point of incubation as well as the first wavelength scan (with 2 nm steps) followed by three additional scans with an interval of 2 minutes. The upper black solid line stands for the last scan at time $t = 6$ minutes. As references, the blue dashed line represents the VP in distilled water, and the red dashed line shows the VP only stored in 0.1 mM H$_2$O$_2$. The grey lines in the graphs serve only for orientation purposes.

No conformational changes were seen by storing VP in distilled water.

4.3. Steady-state Kinetics of the H$_2$O$_2$-dependent Adlerol Conversion by the Crude VP

The reaction velocities of VAld accumulation obtained throughout the steady-state phase showed a hyperbolic shaped course by varying the H$_2$O$_2$ concentration at different fixed adlerol concentrations as depicted in Fig. 7A. Vice versa, same results were obtained by varying the adlerol concentration at several fixed initial H$_2$O$_2$ concentrations (Fig. 8A). Consequently, for each examination procedure two apparent kinetic constants, like the maximum reaction velocity $v_{\text{max app}}$ and the dissociation constant $K_{m app}$, were determined according to the Michaelis-Menten equation type as follows. The index $i$ in Equation (12) stands for the concentration of substrate $i$, such as H$_2$O$_2$ for $i = 1$ and adlerol for $i = 2$.

$$v = \frac{v_{\text{max app}} [S_i]}{K_{m app} + [S_i]}, \quad i = 1, 2 \tag{12}$$

To solve the data-fitting problem in least-square sense the MATLAB® non-linear curve fit function 'lsqcurvefit' (default setting) was used resulting in the kinetic constants listed in Table 2 and 4. For each calculation, initial starting points for $v_{\text{max app}}$ and $K_{m app}$ were estimated from intercepts ($b = 1/v_{\text{max app}}$) and slopes ($m = K_{m app}/v_{\text{max app}}$), respectively, of the reciprocal plots seen in Fig. 7B and 8B by linear regression (general formula: $y(x) = mx + b$). Once, these plots run parallel to each other, ping-pong mechanism exists (Bisswanger, 2008). The plots just partly show parallelism in Fig. 8B, whereas those in Fig. 7B are nearly parallel.

Note: At an initial H$_2$O$_2$ concentration range of 50 µM to 100 µM H$_2$O$_2$ depletion was reached within ca. 3 min, whereas at 15 µM H$_2$O$_2$ the reaction may be already done after ca. 1.5 min at the latest.

Assuming the POX cycle will be not inhibited and irreversible with $k_3 >> k_4$, while $k_4$ is just rate limiting (see simplified two substrate (H$_2$O$_2$ and adlerol (S)) scheme in Fig. 1), the steady-state product (VAld) accumulation can be simply described by Equation (13) (in consideration of Equation (4) and (5); only with VAld as product). A detailed deduction can be found in the study of Rasmussen et al. (1995).

$$\frac{d[VAld]}{dt} = \frac{2k_4[E_1][S_2][S_i]}{k_4[S_i] + [S_i]} \tag{13}$$

with the reaction rate constant:

$$k_4 = k_{cat} \tag{14}$$
Fig. 7. A: VP kinetics for the co-substrate H$_2$O$_2$ at different constant adlerol concentrations as follows: 100 µM (*), 250 µM (+), 500 µM (△), 750 µM (○), 1000 µM (□), and 1500 µM (×). Reactions were conducted in a 200 µL scale in 100 mM sodium tartrate buffer, pH 4.0, at 30°C and an enzyme concentration of ca. 0.3 mg mL$^{-1}$. Computer simulated curves are demonstrated by black solid lines or a dotted black line for c(adlerol) = 1500 µM. B: Double-reciprocal plot of the data shown in A. Due to the enormous similarity of the results obtained at fixed 1000 µM adlerol and 1500 µM, the latter was omitted to provide a better overview. The almost parallel arrangement of the straight solid lines (gained through linear regression ($y = mx + b$)) indicates a ping-pong bi bi mechanism.

Fig. 8. A: VP kinetics for the reducing substrate adlerol at several fixed initial H$_2$O$_2$ concentrations as follows: 15 µM (*), 25 µM (+), 50 µM (△), 75 µM (○), 100 µM (□), 125 µM (×), and 150 µM (×). Reactions were conducted in a 200 µL scale in 100 mM sodium tartrate buffer, pH 4.0, at 30°C and an enzyme concentration of ca. 0.3 mg mL$^{-1}$. Computer simulated curves are demonstrated by black solid lines or a dotted black line for c(H$_2$O$_2$) = 150 µM. B: Double-reciprocal plot of the data shown in A. No parallelism is seen in general. The inset plot show the results of linear regression at initial H$_2$O$_2$ concentrations of 15 µM (*), 25 µM (+) and 125 µM (×) where parallelism can be observed. The black solid lines are gained through linear regression of the general formula $y = mx + b$. 
and the enzyme mass balance

\[
[E_T] = [E^+] + [E^1] + [E^0]
\]  

(15)

The total enzyme concentration \(E_T\) was calculated by assuming 8.6\% of the used lyophilisate are the actual VP amount (in consideration of the determined RZ value of 0.3 compared to the reported 3.5 (Pogni et al., 2005)).

Setting

\[
A = k_4 [S_1]
\]  

(16)

\[
B = \frac{k_4}{k_1} [S_2]
\]  

(17)

(Rasmussen et al., 1995), which is valid and feasible for constant adlerol concentrations (Fig. 7) and rearranging, Equation (13) becomes to Equation (18).

\[
\frac{d[V_{ald}]}{dt} = \frac{A [S_1]}{B + [S_1]}
\]  

(18)

Finally, plots of the coefficients like A vs. B (coefficients were obtained via the same non-linear curve fit approach, as previously discussed using Equation (18)) and A vs. [adlerol] should yield straight lines if the mechanism follows the classical POX cycle and is irreversible (Fig. 1), respectively (Bakovic and Dunford, 1993). As illustrated in Fig. 9, neither plot A nor plot B appears linear. Plot B shows definitely a hyperbola dependence of coefficient A on adlerol, while plot A tends rather towards linearity (but is still not satisfactory following the solid line in plot A of Fig. 9).

By secondary plots of the apparent kinetic parameters \(v_{\text{max}}^{\text{app}}\) and \(K_m^{\text{app}}\) given in Table 2 and 4, both display hyperbolic dependence on adlerol as well as on \(H_2O_2\) concentration (Fig. 10-11), whereas the dissociation constants seem to have a maximum. For adlerol the maximum \(K_m^{\text{app}}\) is ca. 382 \(\mu M\) at a \(H_2O_2\) concentration of 75 \(\mu M\) and for \(H_2O_2\) ca. 13 \(\mu M\) at an adlerol concentration of 750 \(\mu M\) before decreasing to 319 \(\mu M\) and 11 \(\mu M\). Repeated VP reactions at fixed 75 \(\mu M\) and 150 \(\mu M\) \(H_2O_2\) concentrations were done independently and confirmed the findings in Fig. 11.

However, due to the apparent hyperbolic dependence of the kinetic parameters on both substrates a two substrate ping-pong bi bi mechanism will be suggested with formation of binary enzyme-substrate (ES) complexes before irreversible product release occurs (Marangoni, 2003). The generally accepted non-inhibited rate equation, just concerning forward reaction in the absence of product(s), is given through Equation (19) (Bisswanger, 2008; Cornish-Bowden, 2012).

\[
v = \frac{v_{\text{max}} [S_1][S_2]}{K_m^{S_1}[S_1] + [S_2][S_2]}
\]  

(19)

In case \(S_1\) (\(H_2O_2\) concentration) varies and \(S_2\) (adlerol concentration) is treated as constant, which was more realistic for the current study, Equation (19) takes the Michaelis-Menten form seen in Equation (12) (for \(S_i = S_2\)) with the parameters \(v_{\text{max}}^{\text{app}}\) and \(K_m^{\text{app}}\) as follows.

\[
v_{\text{max}}^{\text{app}} = \frac{v_{\text{max}} [S_2]}{K_m^{S_2^{\text{app}}} + [S_2]}
\]  

(20)

\[
K_m^{\text{app}} = \frac{K_m^{S_1} [S_1]}{K_m^{S_1} + [S_2]}
\]  

(21)

By fitting Equation (20) and (21) to the represented data in Fig. 10 using the known non-linear approach, first estimates of the kinetic parameters \(v_{\text{max}}\), \(K_m^{S_1}\) and \(K_m^{S_2}\) were made as recorded in Table 3.
Fig. 10. $v_{\text{max}}^{\text{app}}$ (A), $K_m^{\text{app}}$ (B) dependence on c(adlerol). $v$ and $K$ represent the data listed in Table 2. The black solid line denotes a computational non-linear curve fit as explained in the text by using Equation (20) (A) and Equation (21) (B).

**Table 2.** Apparent kinetic constants $v_{\text{max}}^{\text{app}}$ and $K_m^{\text{app}}$ varying H$_2$O$_2$ at different fixed adlerol concentrations.

| c(adlerol) | $v_{\text{max}}^{\text{app}}$ | $K_m^{\text{app}}$ |
|-----------|-----------------|------------------|
| µM        | µM min$^{-1}$   | µM               |
| 100       | 2.5 ± 0.0       | 4.0 ± 0.6        |
| 250       | 4.5 ± 0.1       | 8.8 ± 1.9        |
| 500       | 6.4 ± 0.1       | 8.1 ± 2.3        |
| 750       | 7.8 ± 0.1       | 13.2 ± 1.8       |
| 1000      | 9.1 ± 0.1       | 10.3 ± 0.8       |
| 1500      | 9.3 ± 0.5       | 10.8 ± 0.9       |

The kinetic constants represent the results of a non-linear curve fit using the mean steady-state reaction velocity of three measurements. All reactions were carried out in 100 mM sodium tartrate buffer (pH 4.0) at T = 30°C.

**Table 3.** Estimates of $v_{\text{max}}$ $K_m^{S_1}$, and $K_m^{S_2}$ for constant adlerol conditions ($S_1$: H$_2$O$_2$, $S_2$: adlerol).

| Kinetic parameter | $v_{\text{max}}$ | $K_m^{S_1}$ | $K_m^{S_2}$ |
|-------------------|-----------------|-------------|-------------|
| µM min$^{-1}$     | µM              | µM          | µM          |
|[S$_2$] = constant | 12.2            | 16.4        | 417         |

Data were calculated by non-linear curve fitting using Equation (20) and (21), respectively.

**4.4. Pre-steady-state Transients**

Prior steady-state (linear reaction rate velocity has been developed) slow transient phenomena in form of a lag phase appeared once H$_2$O$_2$ exceeded a certain concentration (Fig. 12 and 13).

Under the current reaction conditions the threshold will be approximately at c(H$_2$O$_2$) ≥ 50 µM. Based on a constant adlerol concentration (an illustrative example is given in Fig. 12), the lag phase was extended by increasing the initial H$_2$O$_2$ concentration until a maximum is achieved, obviously without affecting the subsequent steady-state velocity $v$ which supports the data in Fig. 7A. Any additional H$_2$O$_2$ increase led to a drop in absorption signal (absorption of the VP-adlerol blank is greater than that of the full reaction mixture as seen in Fig. 14) resulting first in negative reaction rates, apparently of the same initial speed. The negative slope declined with time until it stagnated (a minimum/turning point was reached) before product accumulation could be finally detected. Whereas the time course in Fig. 14 is strongly dependent on the H$_2$O$_2$ load until reaching the turning point, the subsequent product accumulation profile remained unaffected.
Fig. 12. Lag phase formation by increasing the H$_2$O$_2$ amount (50 µM (▲), 75 µM (○), 100 µM (□), 125 µM (◆), and 150 µM (×); inset plot: 15 µM (●), 25 µM (+)) at a fixed adlerol concentration of 500 µM. The corresponding adlerol to H$_2$O$_2$ ratios are 33.3, 20, 10, 6.7, 5, 4, and 3.3. The absorption value (data are already corrected by blank) at a wavelength of 310 nm is plotted as a function of measuring time.

Fig. 13. Lag phase characteristic depending on the adlerol amount (100 µM (■), 250 µM (●), 500 µM (◆), 750 µM (▲), and 1500 µM (+)) at a fixed initial H$_2$O$_2$ concentration of 100 µM. The corresponding adlerol to H$_2$O$_2$ ratios are 1, 2.5, 5, 7.5, and 15. Absorption values (data are already corrected by blank) at a wavelength of 310 nm are plotted as a function of measuring time.

Such events occurred at adlerol to H$_2$O$_2$ ratios ranging from 1.3 (100 µM adlerol and 75 µM H$_2$O$_2$) up to 3.3 (500 µM adlerol and 150 µM H$_2$O$_2$) as long as the adlerol concentration will be maintained at ≤ 500 µM. At concentrations ≥ 750 µM the lag phase could still be monitored. Thus, concerning a fixed initial H$_2$O$_2$ concentration (e.g. 100 µM (Fig. 13)), the duration of the lag period was reduced by elevating the adlerol content. Moreover, the reaction rate rose with adlerol as well.

4.5. Spectral Soret Band Monitoring for Reaction Mechanism Interpretation

In order to clarify the events of several reactions seen above, the soret band behavior was again independently investigated under the same reaction conditions as executed for the kinetic measurements, except that just a fixed adlerol concentration of 1 mM was used. The corresponding results are graphically shown in Fig. 15; the characteristic wavelengths were already listed in Table 1. At λ = 310 nm in Fig. 15, an additional grey line is drawn to follow the VAld accumulation over time, simultaneously. The reaction mixture was monitored by a series of spectroscopic scans (wavelength area between
300 nm and 500 nm, with a step size of 2 nm) made every two minutes over a time period of \( t = 12 \) min (inclusive pre-incubation phase of 7 min) starting from \( t = 0 \) min. The last scan of pre-incubation was performed at \( t = 6 \) min (blue solid line in Fig. 15, all previous scans are omitted). At the time \( t = 7 \) min the reaction was initiated by \( \text{H}_2\text{O}_2 \) addition. The first scan was made one minute later at \( t = 8 \) min (red solid line in Fig. 15).

An additional reaction was conducted at 350 \( \mu \text{M} \) \( \text{H}_2\text{O}_2 \) to simulate a theoretical \( \text{H}_2\text{O}_2 \) overload as reference (Fig. 15F). In such circumstances, preliminary studies using ABTS as reducing substrate showed some degree of inactivation of the used VP (data are not published). Indeed, monitoring VAld, similar results (initial drop in absorption) were received as seen in Fig. 14. Thus, enzyme deactivation may be expected.

![Fig. 15. Monitoring (raw data) of the soret band behavior (scan interval: 2 minutes, step size: 2 nm) after reaction start by adding A: 15 \( \mu \text{M} \), B: 50 \( \mu \text{M} \), C: 75 \( \mu \text{M} \), D: 100 \( \mu \text{M} \), E: 125 \( \mu \text{M} \), and F: 350 \( \mu \text{M} \) \( \text{H}_2\text{O}_2 \) at constant adlerol concentration of 1 mM. The blue solid line represents \( t = 6 \) minutes of pre-incubation while the red solid line is the record of the first minute (or at \( t = 8 \) minutes; pre-incubation plus measurement time) after reaction start. The end of reaction (\( t = 12 \) minutes) is indicated by the upper black solid line, except in A. Here, the lower black solid line depicts the reaction end. Inset plots are zooms of the soret region. The black dotted line acts as reference and shows a spectral scan of a VP sample just incubated in 350 \( \mu \text{M} \) \( \text{H}_2\text{O}_2 \).]
At an initial $H_2O_2$ concentration range of 50 µM to 100 µM, $H_2O_2$ depletion is assumed within ca. three minutes, based on the kinetic measurements above (therefore $t = 10$ min is supposed to be the last scan of reaction), whereas at 15 µM $H_2O_2$ the reaction was already done after 1.5 min at the latest (the scan at $t = 8$ min (red solid line) should only be counted).

In Fig. 15B-F it can be extracted that after $H_2O_2$ addition the soret band (407 nm) experienced a red shift to ca. 420 nm, similar to the spectral scan of a VP sample which was only be incubated in 350 µM $H_2O_2$. That would imply the VP reaction cycle passed through $E_{III}$ before return to $E_0$. Furthermore, the soret band recovered by the end of reaction (around about $H_2O_2$ depletion) at $H_2O_2$ concentrations ≤ 100 µM, while $E_{III}$ remained unchanged above 100 µM $H_2O_2$ (no $H_2O_2$ depletion within the reaction period). Beside the red shift (blue solid line vs. red solid line), a considerable decrease in absorption intensity is seen in Fig. 15E-F, once $H_2O_2$ exceeded 100 µM, which is probably associated with an enzyme inactivation process.

Although, the reaction was almost expired after the first spectrum was recorded in Fig. 15A, the cycle is assumed to follow the normal POX mechanism (Fig. 1) under such low $H_2O_2$ conditions. However, a drop of the soret peak could also be registered at the end of examined period. By reconsidering Fig. 14 (inset plot) in the preceding section in this context, a relative slow but constant decrease in absorption is noticed monitoring the VP-adlerol blank. This phenomenon, which was already mentioned throughout pre-incubation, appeared independently of the adlerol amount in all studies with rates of ≤ 0.002 min⁻¹. The same kind of behavior showed the full reaction assay after achieving $H_2O_2$ depletion. At this point, evidence is taken that the major cause of any observed absorption decay is related to VP instabilities. Instability becomes more significant at low initial $H_2O_2$ concentrations as well as lowered adlerol content (Fig. 15A and to some degree in Fig. 12 and 13 of the previous sub-chapter). VP inactivation and stability studies will be continued in the next section.

### 3.6. VP Inactivation/Deactivation by $H_2O_2$ and the Impact of Adlerol

The influence of $H_2O_2$ concentration on the VP stability was investigated as well as the impact of adlerol. For this purpose, two series of experiments were carried out in a 10-50 mL scale by varying the initial $H_2O_2$ concentration (range: 0-1 mM) at initial adlerol concentrations of 0 mM and 4 mM. The mixtures were continuously stirred with a stirrer speed of ca. 375 rpm. Enzyme stability was determined every three minutes by ABTS assay over a time period of 18 min. The observed deactivation constant $k_{d(obs)}$ was calculated by Equation (27) assuming enzyme deactivation follows first-order kinetics of the general formula given in Equation (22).

$$E \xrightarrow{k_{d(obs)}} E_i$$  \hspace{1cm} (22)

$E$ indicates the active enzyme and $E_i$ the inactivated form. Since enzyme inactivation trended to incompleteness due to an expected $H_2O_2$ consumption the enzyme concentration was modified and Equation (22) becomes to Equation (23).

$$\frac{d[E]}{dt} = k_{d(obs)}[E - E_\infty]$$  \hspace{1cm} (23)

$$E_\infty = E \text{ at } t = \infty$$  \hspace{1cm} (24)

$$[E_0] = [E] + [E_\infty]$$  \hspace{1cm} (25)

Separation of the variables and integration for the boundary conditions

$$E_0 = E \text{ at } t = 0$$  \hspace{1cm} (26)

yields Equation (27).

$$-k_{d(obs)} = \frac{\ln \left( \frac{[E_0] - [E_\infty]}{[E_0] - [E_\infty]} \right)}{t}$$  \hspace{1cm} (27)

In order to receive estimates of the enzyme half-life ($t_{1/2}$) setting:

$$[E_0] - [E_\infty] = 2 ([E] - [E_\infty])$$  \hspace{1cm} (28)

and

$$t = t_{1/2}$$  \hspace{1cm} (29)
After substitution in Equation (27), a subsequent rearrangement to \( t_{1/2} \) yields Equation (30).

\[
t_{1/2} = \frac{\ln(2)}{k_{d(\text{obs})}} \tag{30}
\]

Based on Equation (27), \( k_{d(\text{obs})} \) was obtained from negative slopes of the straight lines generated by plotting the numerator against the denominator \( t \) (primary plots). Secondary plots of \( k_{d(\text{obs})} \) as a function of \( \text{H}_2\text{O}_2 \) concentration in Fig. 16 show a hyperbola dependence. The double-reciprocal plot in Fig. 17 of the same data yields a linear relationship with non-zero intercept. According to Copeland (2000), the VP inactivation is subjected to a two-step mechanism with an initial binding step followed by a slower inactivation procedure. In order to solve the non-linear problem via the 'lsqcurvefit' function in MATLAB®, initial starting points for \( k_i^{\text{app}} \) and \( K_i^{\text{app}} \) were received from the intercept \( (b = 1/k_i^{\text{app}}) \) and the slope \( (m = K_i^{\text{app}}/k_i^{\text{app}}) \), respectively, of the linear regression (red solid line with the general formula \( y = mx + b \), \( x = c(\text{H}_2\text{O}_2) \) and \( y = k_{d(\text{obs})} \)) made in Fig. 17. The results are listed in Table 5. In the presence of adlerol \( K_i^{\text{app}} \) increased, while \( k_i^{\text{app}} \) remained almost constant round about 0.3 min\(^{-1}\) comparing the red solid line (0 mM adlerol) with the black one (4 mM adlerol) in Fig. 16 and 17. Consequently, the inactivation mechanism was competitive inhibited by adlerol (denoted as \( S_2 \)) which could be expressed via Equation (32). This equation also allows a first estimation of the adlerol dissociation constant \( K_m^{\text{app}} \). For this purpose, the adlerol concentration was again considered to be constant. Initial values for \( k_i^{\text{app}} \) and \( K_i^{\text{app}} \) were transferred from previous calculations (Equation (31)). For \( K_m^{\text{app}} \) 0.417 mM was specified initially based on the results in the sections above. Finally, the kinetic parameters were adjusted by non-linear curve fitting of both data sets (with and without adlerol) offering a reevaluation of \( k_i^{\text{app}} \) and \( K_i^{\text{app}} \) simultaneously. The data are also stored in Table 5.

\[
k_{d(\text{obs})} = \frac{k_i^{\text{app}}[S_i]}{K_i^{\text{app}}[S_i] + [S_i]} \tag{32}
\]

The ratio \( k_i^{\text{app}}/K_i^{\text{app}} \) reflects a second-order rate constant and is a measure of the inhibitory potency of an irreversible inhibitor (Copeland, 2000), such as \( \text{H}_2\text{O}_2 \) in this case. The obtained value was 25 M\(^{-1}\) s\(^{-1}\).

Due to the inactivation characteristics in the presence of adlerol, Fig. 16 can be clearly extracted that adlerol improved enzyme stability, in general. However, the half-life (inset plot of Fig. 16) decreased relative fast by an elevation of \( \text{H}_2\text{O}_2 \) up to 100 \( \mu \)M and strongly slowed down immediately afterwards.

5. DISCUSSION

The optimal pH of the VP being examined for the Mn-independent adlerol degradation was 3.5-4.0. With the same VP, but subsequently purified, an activity maximum at pH 3.0 (50 mM sodium tratrate buffer; 20°C) was recorded for VA conversion (Liers et al., 2010). Based on Table 6, the pH optimum of ligninolytic enzymes (VP, LiP) varies between 3.0 and 4.0. In case of a polycyclic aromatic substrate a Mn-independent pH optimum was found at 4.0 (Wang et al., 2003). In previous studies (data are still unpublished), pH values of \( \leq 3.5 \) (50 mM sodium tratrate buffer) showed a higher impact on enzyme stability than temperature changes within 25-30°C. Moreover, the pH optimum for lignin degradation by ligninolytic cultures from \( P. \) chrysosporium is 4.5 (Schoemaker, 1990).
Despite the fact, the used high concentrated buffer system seemed to slightly interfere the soret peak morphology, it was not possible to distinguish between $E_{II}$ and $E_{III}$, since differentiation is difficult anyway (maxima are close together as seen in Table 1). Thus, a comparison was made with references, for instance, where VP was stored in distilled water only (reference for $E_{0}$) or just in $H_2O$ (reference for $E_{III}$ formation).

Indeed, conformational changes (kind of a burst phase, Fig. 5) could be recognized throughout enzyme pre-incubation with adlerol. For some reason, it was found that the VP was in its oxyform $E_{III}$ before it was brought up to its ground ferric state $E_{0}$. This process was strongly affected by adlerol concentration (Fig. 5). Consequently, the question arises if this phenomenon is related to binary enzyme-substrate (ES) complex formation and stabilization processes, or does $E_{III}$ convert adlerol. There are no official statements regarding complex formation of ligninolytic POX with aromatic substrates as well as analogue by today, since the enzyme is not designed for direct contact with its centred heme group (Ruiz-Dueñas et al., 2009a).

Contrary, an ABTS conversion study by HRP indicated a ter bi ping-pong mechanism (Cleland, 1963) with one ternary complex formation ($E$-$H_2O_2$-ABTS, while $E$-$H_2O_2$ was defined as $E^0$) prior HRP was transferred to its transition state $E_{II}$ and first product release occurred (Child and Bradshley, 1975).

In general, $H_2O_2$ and reducing substrate do not bind on the same “active site” of the enzyme. Two separate access channels are identified in the literature. The first one is conserved in all POX and quasi reserved for $H_2O_2$. Table 5. Determined inactivation constants $k_{iapp}$ and $K_{iapp}$ with simultaneous $K_m$ estimation.

| c(adlerol) µM | $k_{iapp}$ min$^{-1}$ | $K_{iapp}$ µM | $k_{iapp}/K_{iapp}$ M$^{-1}$ s$^{-1}$ | $K_{m}$ µM |
|--------------|-------------------|-------------|---------------------------------|---------|
| 0            | 0.25              | 145         | 28.7$^{1)}$                      | n.a.$^{2)}$ |
| 4000         | 0.28              | 188         | 24.8$^{1)}$                      | 2,506   |

The kinetic constants represent the results of a non-linear curve fit using the mean observed deactivation constant $k_{d(obs)}$ of two measurements.

1) “best measure for inhibitory potency for an irreversible inhibitor is the second-order rate constant obtained from the ratio $k_{iapp}/K_{iapp}$” (Copeland, 2002)

2) not applicable
(direct contact to the enzyme’s heme group), the second serves for reducing substrate (Ruiz-Dueñas et al., 2009a). Due to the design of the second channel the enzyme substrate contact takes place at the enzyme’s surface via a tryptophan component (in case of VP from B. adusta) which is supposed to be responsible for substrate oxidation, in its radical form (Pogni et al., 2005; Ruiz-Dueñas et al., 2009a).

However, based on the experimental data a conversion of adlerol by EIII would automatically imply the inability of E0 to degrade such a lignin model dimer as it is displayed through pathway 15 in Fig. 3. A possible E1 formation due to reaction of EIII with adlerol (see also pathway 8 in Fig. 3) causing the said enzyme transformation could not be determined accurately (Fig. 6). Even it would be true, further conversion resulting in product (VAld) accumulation is expected as consequence, provided that E1 will be the defined active intermediate. Since it was found that adlerol was not further decomposed to VAld and saturation appeared, this theory is more unlikely.

VP kinetics could be sufficiently described by the well-known Michaleis-Menten equation. Both, H2O2 and adlerol had promotional effects on reaction velocity v in general. Nevertheless, pre-steady-state transients were recognized characterized by a H2O2-dependent lag phase in the time course of VAld accumulation. Usually, transient states are more expected to be fast (ms time scale) and may just be detectable throughout rapid-reaction techniques (Eisenthal and Danson, 2002). Indeed, the said lag phase was slow enough for sensing and appeared at H2O2 concentrations ≥ 50 µM under current reaction conditions. On the basis of the red shift of the soret band (Fig. 15), it will be presumed that the main cause of this phenomenon must have been an accumulation of EIII via EII reaction with H2O2. Inhibitory effects of the co-substrate H2O2 in such a way are known to slow down the reaction (Cai and Tien, 1992; Nicell et al., 1993; Vlasits et al., 2010).

Goodwin et al. (1994) obtained similar results studying the LiP catalyzed oxygen consumption with VA, and in presence of EDTA and oxalate as well. The O2 reduction rate appeared linear at low H2O2 concentration of 30 µM, whereas at values within the range of 90 µM and 120 µM lag phases could be noticed for the same reason (EIII formation).

If EIII can be transferred back to EII was reviewed by pre-incubating the VP in H2O2 (EIII formation was expected), under the known conditions, followed by adlerol addition for initiating the reaction. After a lag phase VAld production gradually restarted (Fig. 18) most likely due to EIII disappearance. It is conceivable that several enzyme molecules were still active (since no response in absorption signal could be detected after complete enzyme inactivation) to decompose adlerol along with certain products which served as substrate for EIII. This requires H2O2 availability, though only in small amounts. The theory is based on studies examining the conversion of EIII of a LiP (LiP-EIII) from P. chrysosporium and VA besides 1,2,4,5-tetra-methoxy-benzene as substrates (Barr and Aust, 1994). The researcher have been shown LiP-EIII disappearance due to reaction with cation radical products to LiP-EII resulting in a reaction rate increase with time. If EIII of the used VP will be reactivated by an adlerol cation radical or a veratryl alcohol radical (VA’), this is questionable. It is proven that LiP-EIII conversion to LiP-EI cannot occur via reaction with VA’, but with a veratryl alcohol cation radical (VA’+) (Cai and Tien, 1992; Chung and Aust, 1995).
Furthermore, VA’ is supposed to rapidly react with O$_2$ to VAld under aerobic conditions (Schoemaker et al., 1994b). Since the kinetics were conducted in O$_2$ atmosphere, VA’ as reductant for E$^{III}$ can be excluded. Aerobic conditions may be further promoted due to O$_2$ generation as a result of E$^{III}$ formation (Nakajima and Yamazaki, 1987; Goodwin et al., 1994). On the other hand, cation radical products are also limited in their lifetime. For VA’ a half-life of 40 ms was recorded (Harvey et al., 1993). Finally, the radical decay is dependent on the nature of its substitutes (e.g. OCH$_3$ (OMe) groups in Fig. 2) favor formation and stabilization) (Wong, 2009).

A conversion of E$^{III}$ back to E$^{II}$ by oxidation of an adlerol cation radical would imply less VAld accumulation, since one adlerol molecule will be only consumed throughout one reaction cycle. At least as long as the reaction passes the E$^{III}$ intermediate. Thus, the reaction with adlerol has to be reconsidered at this point. At the end of the pre-incubation period with H$_2$O$_2$, E$^{III}$ is the perdominant intermediate (supported by soret band and by Barr and Aust (1994)) and a certain portion of enzyme is merely reversibly inhibited. That means, E$^{III}$ may be the only reactant for adlerol. For evidence, further investigations have to be carried out.

When H$_2$O$_2$ concentration was additionally increased and exceeded a certain limit (depending on the available adlerol content) noticeable rapid drop in the absorption signal was the consequence (Fig. 14 and 15E-F). Such H$_2$O$_2$ concentrations were obviously sufficient to cause some degree of irreversible enzyme deactivation. Due to the fact that H$_2$O$_2$ will be consumed throughout the process the drop declined with time. Once acceptable catalytically conditions were restored, accumulation of VAld was again measurable. This could be an explanation for the gradually decline of the apparent $K_m^{app}$ at H$_2$O$_2$ values ≥ 100 µM in Fig. 11B. Since H$_2$O$_2$ degraded faster than adlerol (mechanism-based), the enzyme:adlerol:H$_2$O$_2$ ratio was probably modified in such a way that the reaction rate $v$ remained almost constant as seen in Fig. 7. Nevertheless, enzyme deactivation cannot be fully excluded even in the presence of sufficient adlerol content as well as by recovering E$^{II}$ at low H$_2$O$_2$ concentrations as it was seen in Fig. 15A-D. Although the current deactivation studies have demonstrated protective properties of adlerol, the enzyme activity/stability still undergoes a considerable impact by H$_2$O$_2$ concentrations up to 100 µM at clearly increased adlerol amounts of 4 mM (Fig. 16 and 17). In this context, Aitken and Irvine (1989) investigated deactivation kinetics of a LiP (and a MnP) from P. chrysosporium. Both working groups concluded that the lag phases, caused by E$^{III}$ formation, are additionally associated with enzyme deactivation via E$^{III}$. It was found that the drop in the restored soret peak was proportional to loss in enzyme activity (Hu et al., 1993).

Based on the soret band behavior, deactivation of the crude VP also appeared mainly through compound E$^{III}$, which will be promoted by the used acidic pH. A quick check showed that E$^{III}$ formation did not occur when the VP under study was stored in distilled water, instead of the known buffer system, spiked with the same H$_2$O$_2$ amount. An enhanced susceptibility of a VP from Pleurotus pulmonarius for H$_2$O$_2$ with decreasing pH was also confirmed by Böckle et al. (1999).

Liers et al. (2010) examined VP (B. adusta) stability under acidic conditions (50 mM sodium tartrate buffer, pH 2.5) at 20°C. 70% loss in activity was recorded after 1 h of incubation. In the current work a half-life time of 52 min was found in the presence of adlerol at 30°C and 15 min once 0.1 mM H$_2$O$_2$ (initial concentration) was added, while the stability was halved in the absence of adlerol. A half-life time of just 1.3 min has even been reported in presence of 1 mM H$_2$O$_2$ (no substrate; 60 mM phosphate buffer, pH 6.1) (Valderrama and Vazquez-Duhalt, 2005). In this study, VP deactivation mechanism by H$_2$O$_2$ represents a time-dependent irreversible inhibition and is competitive inhibited in the presence of adlerol. The deactivation kinetic could be described satisfactorily by the model in Equation (32) assuming constant adlerol conditions. From estimates of $k_i^{app}$ and $K_i^{app}$ an average second-order rate constant of ca. 25 M$^{-1}$ s$^{-1}$ was calculated. 16 M$^{-1}$ s$^{-1}$ (pH 3.0; 23°C) were measured for the H$_2$O$_2$-dependent inactivation process via E$^{III}$ of a LiP (Aitken and Irvine, 1989). Furthermore, an additional estimation for the dissociation constant $K_m^{S2}$ of 2,506 µM was obtained indicating high substrate affinity. In comparison to the steady-state kinetics a $K_m^{S2}$ value of just 417 µM (Table 3) was determined at constant adlerol conditions. On the basis of the argumentation above, the estimated $K_m^{S2}$ and the apparent steady-state kinetic parameters may differ from the true values, since a portion of enzyme and H$_2$O$_2$ was partly lost when linear reaction velocities could be determined. Thus, the initial planned concentrations were not fully met. In such circumstances, obtained reaction velocities are underestimated (Kulmacz, 1986).
As pointed out at the beginning, kinetic constants for adlerol are seldom in the literature; none were found for VP. Instead, sufficient data are available for the reducing substrate VA (just for orientation purposes) and also a few for LiP and adlerol conversion. In Table 6 several results from literature are listed for VP (Mn²⁺-independent) and LiP. As it can be seen, $K_m$ values > 3,000 µM are quite conceivable for VPs, also in conjunction with VA, whereas those for LiP are considerably lower. According to Liers et al. (2010), VP is less active toward non-phenolic lignin model compounds than LiP. In addition, the question was raised as whether for ligninolytic peroxidases a great substrate excess (e.g. 25 mM) has to be required in order to reach maximal reaction rates (German et al., 2011).

Finally, based on the current observations, the crude VP reaction mechanism for adlerol degradation will be assumed as summarized in Fig. 19 for the next investigations, such as developing a $H_2O_2$ feeding strategy.

**Fig. 19.** Supposed $H_2O_2$-dependent reaction mechanism of the crude VP from *B. adusta* for adlerol degradation. $E^0$-$E^{III}$ are enzyme intermediates. The symbols $S$ and $S^+$ stand for an appropriate substrate (e.g. adlerol) and its corresponding radical cation, respectively. $O_2^-$ denotes superoxide radical anions. Pathway 1-3 depicts the usual POX reaction cycle with the reversible steps described by Equation (a) and (b) (Ruiz-Dueñas et al., 2009b). The cycle will be initiated by $H_2O_2$ (pathway 1). Pathway 4, 5, and 7 show important side reactions with $H_2O_2$ which competes with pathway 6. Enzyme deactivation ($E_i$: inactivated enzyme) is found to occur via $E^{III}$ as sketched in pathway 7. Pathway 8 illustrates a spontaneous unimolecular decay of $E^{III}$.
Table 6. Compilation of some kinetic investigation results for H$_2$O$_2$ and different monomeric and polymeric aromatic substrates.

| Peroxidase | Substrate S | $k_{cat}$ | $V_{max}$ | $K_m$ | Initial reaction conditions | Reference |
|------------|-------------|---------|--------|-----|-----------------------------|-----------|
| VP (Bjerkandera adusta) | H$_2$O$_2$ | n.a. | n.a. | 200 | pH: 4.0 / T: 25°C / 50 mM malonate buffer / reducing substrate: PAH$^1$ | Wang et al., 2003 |
| | | | | 23,800 | 0.1 mM H$_2$O$_2$ | |
| | PAH$^1$ | 2.4$^2$ | n.a. | | | |
| VP (Bjerkandera species) | H$_2$O$_2$ | n.a. | 28 | 182 | pH: 3.0 / RT / 0.5 mM VA / ca. 0.1 mM sodium tartrate buffer | Moreira et al., 2006 |
| | VA | n.a. | 13 | 1,500 | pH: 3.0 / ca. 320 µM H$_2$O$_2$ | |
| | VA | n.a. | 83 | 3,670 | pH: 5.0 | |
| VP (Bjerkandera species strain BOS55) | VA | 1.4-2.8 | 7.6-15.6$^3$ | 116-534 | pH: 3.0 and 4.5 / 0.1 mM H$_2$O$_2$ / 50 mM succinate buffer / [E]: 4 mg L$^{-1}$ | Mester and Field, 1998 |
| | H$_2$O$_2$ | n.a. | n.a. | 2-3 | pH: 3.0 / 2 mM VA / 0.1 mM sodium tartrate buffer | Camarero et al., 1999 |
| | VA | n.a. | n.a. | 3,500 | pH: 3.0 / 0.4 mM H$_2$O$_2$ / 0.1 mM sodium tartrate buffer | |
| | VA | n.a. | 45-95 | 3,000-3,500 | pH: 3.0 / 0.1 mM H$_2$O$_2$ / 0.1 mM sodium tartrate buffer | Martinez et al., 1996 |
| VP (Pleurotus eryngii) | VA | 7.1 | 17.0$^4$ | 149 | pH: 3.0 / T: 25°C / 0.2 mM H$_2$O$_2$ / 0.05-2.5 mM S / 125 mM tartrate / [E]: 1.8 µM | Cho et al., 2010 |
| | H$_2$O$_2$ | 3.7 | 8.9$^4$ | 11 | pH: 3.0 / T: 25°C / 1-50 µM H$_2$O$_2$ / 1 mM VA / [E]: 40 nM | Lundell et al., 1993b |
| LiP (Phlebia radiate) | VA | 4.7 | 11.3$^4$ | 192 | similar to VA above | |
| | H$_2$O$_2$ | 3.3 | 5.6$^4$ | 55 | pH: 3.0 / T: 37°C / 0.2 mM H$_2$O$_2$ / 0.1 mM sodium tartrate buffer / 0.1% Tween 80 / 5 µg mL$^{-1}$ protein | Tien and Kirk, 1984 |
| | adlerol | 4.7 | 11.3$^4$ | 192 | similar to VA above | |
| | dimeric non-phenolic LMC$^5$ | n.a. | n.a. | 55 | | |
| LiP (Phanerochaete chrysosporium) | β-O-4 (adlerol) | 0.5 | 54$^4$ | 160 | pH: 3.5 / T: 25°C / 50 µM H$_2$O$_2$ / 0.05-2.5 mM S / 125 mM tartrate / [E]: 1.8 µM | |

1) PAH: polycyclic aromatic hydrocarbon,  
2) original data was 145 min$^{-1}$,  
3) original data were 1.9-3.9 U mg$^{-1}$,  
4) subsequently calculated based on given data assuming $v_{max} = k_{cat} [E]$,  
5) 1,2-bis-(3-methoxy-4-methoxyphenyl)-propane-1,3-diol
It is suggested that the normal VP reaction cycle (without \( E^{III} \) formation) involves reversible reaction steps. Despite a further review regarding applicability of the kinetic models described through Equation (13) or (19) as well. An approval is given by Ruiz-Dueñas et al. (2009b) who investigated transient state kinetics of a recombinant native VP (obtained by \textit{E. coli}) with the substrate VA (Equation (6)-(7)). \( E^I \) formation via two-electron oxidation of \( E^0 \) by \( H_2O_2 \) has long been understood (Dunford, 2010). Moreover, it will be supposed that \( E^{III} \) is directly reduced by adlerol (assumption is general accepted, e.g. for VA oxidation (Ruiz-Dueñas et al., 2009b)). However, the evidence for this assumption is still pending.

Deactivation of the crude VP is supposed to occur as described by pathway 7 (Fig. 19). Nevertheless, activity loss via spontaneous unimolecular decay of \( E^I \) (is unstable (Dunford, 2010); is omitted in Fig. 19) and \( E^{III} \) to \( E^{II} \) and \( E^0 \), respectively, have to be taken into account for mathematical modeling and process simulation. The same probably applies for side reaction of \( E^I \) with \( H_2O_2 \) to \( E^{III} \) (pathway 4, Fig. 19) or even directly to \( E^{III} \) (Vlasits et al., 2010), in case \( H_2O_2 \) exceeds a certain amount.

6. CONCLUSION

“Due to the complexity of the degradation reactions of \( \beta-O-4 \) compounds it is difficult to find solid experimental support” (Lundell et al., 1993a). In addition, definite conclusions are generally hampered, since a crude enzyme was used. However, focusing bioprocess engineering, e.g. implementation of online monitoring systems along with developing feeding strategies, important insights could be gained into the reaction mechanisms as follows.

The crude VP from \textit{B. adusta} showed saturation kinetics (described by Michaelis-Menten equation) for adlerol degradation with relative sensitive response to \( H_2O_2 \). This was characterized by slow transient states in form of a lag phase most likely caused by \( E^{III} \) formation via \( E^I \) reaction with \( H_2O_2 \) prior steady state. Furthermore, \( E^{III} \) could be (partly) converted back to \( E^0 \) for restarting a catalytic cycle. Whether that was triggered by adlerol or its cation radical product still has to be verified.

The VP deactivation by \( H_2O_2 \) followed a time-dependent irreversible mechanism and occurred mainly through \( E^{III} \). Deactivation was diminished in presence of adlerol indicating protective properties of the substrate. Hence, evidence is taken that a competition occurs between \( H_2O_2 \) and adlerol or its radical cation products, respectively, for reactions with \( E^{III} \) as well as \( E^{II} \).

Regardless the proven catalytic activity of \( E^{III} \), it is recommended to maintain \( H_2O_2 \) concentrations below 50 \( \mu M \) at an adlerol to \( H_2O_2 \) ratio of at least 15:1 in order to minimize enzyme reactions as well as enzyme losses via \( E^{III} \). From the bioprocess engineering point of view, this implies relatively slow \( H_2O_2 \) feeding rates, or in other words, a fine tuning of \( H_2O_2 \) concentration concerning fed-batch and continuous operational modes in bioreactors. Indeed, low \textit{in vitro} \( H_2O_2 \) concentrations are assumed (Hammel et al., 1993; Böckle et al., 1999), since \( H_2O_2 \) is obviously assimilated by the fungal mycelium avoiding unfavorable high concentrations \textit{in vivo} (Böckle et al., 1999). In addition, the reducing substrate amount and the enzyme needs to be balanced as well.

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