



HAL
open science

Identification of the molecular determinants driving the substrate specificity of fungal lytic polysaccharide monooxygenases (LPMOs)

Kristian Frandsen, Mireille Haon, Sacha Grisel, Bernard Henrissat, Leila Lo Leggio, Jean-Guy Berrin

► To cite this version:

Kristian Frandsen, Mireille Haon, Sacha Grisel, Bernard Henrissat, Leila Lo Leggio, et al.. Identification of the molecular determinants driving the substrate specificity of fungal lytic polysaccharide monooxygenases (LPMOs). *Journal of Biological Chemistry*, 2021, 296, pp.100086. 10.1074/jbc.RA120.015545 . hal-03126082

HAL Id: hal-03126082

<https://hal.inrae.fr/hal-03126082v1>

Submitted on 11 Jul 2024

HAL is a multi-disciplinary open access archive for the deposit and dissemination of scientific research documents, whether they are published or not. The documents may come from teaching and research institutions in France or abroad, or from public or private research centers.

L'archive ouverte pluridisciplinaire **HAL**, est destinée au dépôt et à la diffusion de documents scientifiques de niveau recherche, publiés ou non, émanant des établissements d'enseignement et de recherche français ou étrangers, des laboratoires publics ou privés.



Distributed under a Creative Commons Attribution 4.0 International License



Identification of the molecular determinants driving the substrate specificity of fungal lytic polysaccharide monoxygenases (LPMOs)

Received for publication, August 7, 2020, and in revised form, November 16, 2020. Published, Papers in Press, November 16, 2020.

<https://doi.org/10.1074/jbc.RA120.015545>

Kristian E. H. Frandsen^{1,2}, Mireille Haon¹, Sacha Grisel¹, Bernard Henrissat^{3,4,5}, Leila Lo Leggio², and Jean-Guy Berrin^{1,*}

From the ¹INRAE, Aix-Marseille University, Polytech Marseille, UMR1163 BBF, Marseille, France; ²Department of Chemistry, University of Copenhagen, Copenhagen, Denmark; ³Architecture et Fonction des Macromolécules Biologiques (AFMB), CNRS, Aix-Marseille Université, Marseille, France; ⁴INRAE, USC1408 Architecture et Fonction des Macromolécules Biologiques (AFMB), Marseille, France; and ⁵Department of Biological Sciences, King Abdulaziz University, Jeddah, Saudi Arabia

Edited by Gerald Hart

Understanding enzymatic breakdown of plant biomass is crucial to develop nature-inspired biotechnological processes. Lytic polysaccharide monoxygenases (LPMOs) are microbial enzymes secreted by fungal saprotrophs involved in carbon recycling. LPMOs modify biomass by oxidatively cleaving polysaccharides, thereby enhancing the efficiency of glycoside hydrolases. Fungal AA9 LPMOs are active on cellulose, but some members also display activity on hemicelluloses and/or oligosaccharides. Although the active site subsites are well defined for a few model LPMOs, the molecular determinants driving broad substrate specificity are still not easily predictable. Based on bioinformatic clustering and sequence alignments, we selected seven fungal AA9 LPMOs that differ in the amino-acid residues constituting their subsites. Investigation of their substrate specificities revealed that all these LPMOs are active on cellulose and cello-oligosaccharides, as well as plant cell wall-derived hemicellulosic polysaccharides, and carry out C4 oxidative cleavage. The product profiles from cello-oligosaccharide degradation suggest that the subtle differences in amino-acid sequence within the substrate-binding loop regions lead to different preferred binding modes. Our functional analyses allowed us to probe the molecular determinants of substrate binding within two AA9 LPMO subclusters. Many wood-degrading fungal species rich in AA9 genes have at least one AA9 enzyme with structural loop features that allow recognition of short β -(1,4)-linked glucan chains. Time-course monitoring of these AA9 LPMOs on cello-oligosaccharides also provides a useful model system for mechanistic studies of LPMO catalysis. These results are valuable for the understanding of LPMO contribution to wood decaying process in nature and for the development of sustainable biorefineries.

Efficient conversion of plant biomass for the production of biofuels and other sustainable bioproducts and materials is considered largely dependent on the key enzymes lytic

polysaccharide monoxygenases (LPMOs) (1, 2). LPMOs are copper-dependent oxidoreductases that use molecular oxygen or hydrogen peroxide as cosubstrate to cleave polysaccharides (e.g. cellulose, hemicellulose, chitin, starch) through an oxidative mechanism (3–5). LPMOs exist either as single-domain enzymes or appended to other protein domains, for example, carbohydrate-binding modules (CBMs) (6, 7), directing the enzymes to their target substrate (8–10). To date, LPMOs are grouped into seven families in the CAZy database (11), namely the auxiliary activity (AA) families AA9-AA11 and AA13-AA16 (7), in which members have been found mainly in bacteria and filamentous fungi (12–17), but also recently in arthropod and fern species (18, 19). In all LPMOs studied to date, the active-site redox center is made up of the strictly conserved histidine brace (His-brace) motif, which consists of the N-terminal His and a second His involved in the coordination of one copper atom (20, 21). The AA9 family also contains some members without any LPMO activity, lacking both the His-brace and copper, while features implicated in carbohydrate binding have been maintained (22).

Genes encoding AA9 LPMO enzymes are found in high numbers in fungal saprotrophs. For instance, species of the *Lentinus*, *Phanerochaete*, *Neurospora*, and *Podospora* genera display between 14 and 33 AA9 genes (<https://mycocosm.jgi.doe.gov/mycocosm/home>); however, the reason for this gene multiplicity is unknown. Structurally, these AA9 isoforms share a central β -sandwich core, but the surface-exposed structural features around the His-brace vary as reviewed (2, 21, 23). The LPMO structure from *Thermoascus aurantiacus* (TaAA9A) (20) revealed a Cu-bound His-brace on a flat extended surface near a conserved Tyr, in the C-terminal (LC) loop, putatively involved in substrate binding. AA9 LPMOs were initially assumed to specifically target crystalline regions of polysaccharides, but later, the activity was also demonstrated on a range of hemicellulose polysaccharides such as xyloglucan (XG), xylan, glucomannan (GM), and mixed-linkage β -glucans (MLGs), and also on oligosaccharides (17, 24–31). The AA9 LPMO from

This article contains [supporting information](#).

* For correspondence: Jean-Guy Berrin, jean-guy.berrin@inrae.fr.



Investigation of seven fungal AA9 LPMOs

Neurospora crassa (NcAA9C) is active on both cello-oligosaccharides and hemicellulosic substrates, suggesting that the enzyme recognizes small stretches of β -1,4-linked glucosyl units (24, 25). Borisova *et al.* (32) determined the structure of NcAA9C and noticed three consecutive Asn residues in the L2 loop and an insertion in the L3 loop of NcAA9C structure compared with other AA9 LPMOs (such as TaAA9A) acting on insoluble cellulosic substrates (Fig. 1 and Fig. S1). Around the same time, we found that an AA9 LPMO from *Podospora anserina* (PaAA9H) with an L3 loop similar to that of NcAA9C showed activity on cello-oligosaccharides as well as XG and GM (29). Shortly after, we determined the structures of two AA9 LPMOs from *Lentinus similis* (LsAA9A) and *Collariella virescens* (CvAA9A, previously *Chaetomium virescens*), which both belong to the phylogenetic cluster C38 (see Results) and display activity on cello-oligosaccharides and on XG, MLG, and GM (28, 33). The L3 loops in LsAA9A and CvAA9A are slightly shorter than those in NcAA9C and PaAA9H; however, their L3 loops are all extended compared with those of TaAA9A, which is not active on oligosaccharides. Interestingly, truncation of the L3 loop in NcAA9C abolishes XG activity (34), and for a closely related (about 60% sequence identity) AA9 LPMO from *Chaetomium thermophilum* (denoted as CtPMO1 [35]), a His-to-Ala mutation in the L3 loop (equivalent to His66 in LsAA9A, see below) has been shown to abolish celloheptaose activity.

The first LPMO–carbohydrate complex structures determined were LsAA9A in complex with cello-oligosaccharides (27, 36). Later, LsAA9A was determined in complex with oligosaccharides derived from hemicelluloses (xylan, GM, and MLG) (28). The LsAA9 complex structures confirmed that the oligosaccharides were bound by one Asn in the L2 loop, polar amino-acid residues in the L3 loop, and the conserved Tyr platform (Tyr 203) in the LC loop but also via charged residues in the L8 loop (after notation [37]). Subsites from -4 to $+2$ were defined based on the LsAA9–cellohexaose (cell6) complex. The negative subsites (-4 to -1) were formed by the platform Tyr203 in the LC loop as well as Glu148, Asp150, and Arg159 in the L8 loop, whereas the positive subsites ($+1$ to $+2$) were formed by Asn28 in the L2 loop and His66 and Asn67 in the L3 loop. Interestingly, the structures also revealed an uncommon lone pair– π aromatic interaction between the pyranose ring O5 and the imidazole of the N-terminal His of the His brace (27, 36). Very recently, crystallographic cello-oligosaccharide complexes were also obtained with CvAA9A and showed broadly the same interactions as LsAA9A (Tandrup *et al.*, submitted; 6YDC, 6YDD, and 6YDE) (33).

Although the substrate-binding determinants and active-site subsites are well defined for LsAA9A, the molecular determinants of substrate specificity are still not easily predictable for other AA9 LPMOs. In this study, we focused on seven LsAA9A-related enzymes from fungal saprotrophs to provide further insights into the main molecular determinants driving LPMO activity toward cello-oligosaccharides and hemicelluloses.

Results

Selection and production of seven AA9 LPMOs

The selection of AA9 sequences was carried out bioinformatically by searching for sequences with at least 50% sequence identity to LsAA9A in the CAZy (<http://www.cazy.org/>) and JGI MycoCosm (<https://mycoCosm.jgi.doe.gov/mycoCosm/home>) databases. We retrieved 100 sequences mainly from basidiomycete species belonging to the Agaricomycetes class but also from ascomycete species belonging to the *Gyromitra* and *Aspergillus* genera (Peizizomycetes or Eurotiomycetes class, respectively). The vast majority of these species hold around 20 or more AA9 LPMO genes. Most of these fungi appeared only once in the list, whereas some appeared twice (*Gyromitra esculenta*, *Armillaria ostoyae*, *Crepidotus variabilis*, *Botryobasidium botryosum*, *Schizophyllum commune*, *Volvariella volvacea*) or even three or four times (*Crucibulum laeve* and *Coprinellus pellucidus*). It means that these fungi display at least one LsAA9A-related LPMO that may have a dedicated biological function.

LsAA9A belongs to C38, and the 100 LsAA9A-related sequences are classified within either subcluster C29 or C38, as previously defined by Lenfant *et al.* (6) based on the sequence conservation of AA9 N-terminal halves, that is, the N-terminal part of the sequences bordered by the residues forming the His brace. Of the 100 sequences, seven sequences were finally selected based on the composition of their putative substrate-interacting loops, L2, L3, L8, and LC (Fig. 1 and Fig. S1). The five AA9 sequences belonging to subcluster C38 originate from the basidiomycetes *Phanerochaete chrysosporium* (PchAA9E JGI protein ID 2934397), *Phanerochaete carnosa* (PcaAA9A 261285), *Bjerkandera adusta* (BaAA9A 353490), *Armillaria gallica* (AgAA9A 500811), and *Schizophyllum commune* (ScAA9A 2617723) and share between 63 and 76% sequence identity with LsAA9A (Table 1). Two other AA9s belong to subcluster C29 and originate from the ascomycetes *Aspergillus oryzae* (AoAA9A UniProt Q2US83) and *Aspergillus fumigatus* (AfaAA9C GenBank EAL85444.1) (Fig. 1, Tables 1, and S1). AoAA9A and AfaAA9C naturally harbor a CBM1 module, and their catalytic AA9 domains share 53 and 55% sequence identity with LsAA9A, respectively (Table 1). Some of the candidates chosen in this study originate from well-known fungal species for which other AA9 LPMOs have already been characterized, for example, *P. chrysosporium* PcGH61D (38, 39) and *A. fumigatus* AfaAA9B (40). However, no activity on oligosaccharides or hemicelluloses has been reported for any AA9 enzymes from these organisms.

Compared with LsAA9A, the sequences of PchAA9, PcaAA9, and BaAA9 have a conservative substitution of the substrate-interacting Glu148 to a Gln near the -2 subsite, which we will refer to as E148Q. From here on, we will use this notation to refer to natural variations of the selected AA9 LPMOs compared with LsAA9A (note that this notation does not refer to mutant variants). The sequence of BaAA9 has an additional N67D substitution potentially affecting the $+2$ subsite. The sequences of AgAA9, ScAA9A, AoAA9, and AfaAA9 also have the N67D substitution together with other ones near the negative

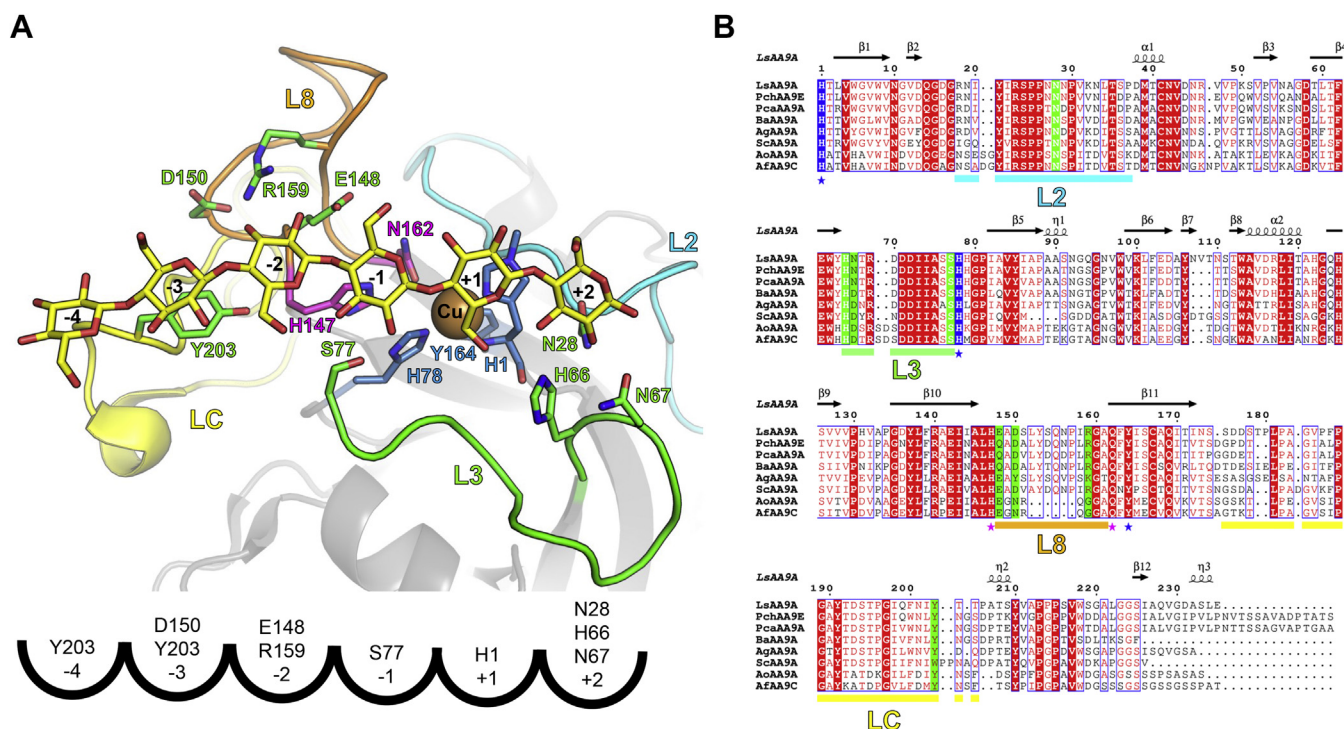


Figure 1. Close-up view of LsAA9A active site and sequence alignment of the AA9 LPMOs studied highlighting the residues putatively involved in substrate binding. A, LsAA9A-cell6 complex (PDB 5AC1) highlighting the subsites -4 to +2 and below schematic representation with residues contributing to substrate binding by hydrogen bonding or aromatic stacking or van der Waals interactions in each subsite. These residues are shown in sticks along with the residues involved in copper coordination (including the secondary sphere). The structural loop elements are colored as in panel B. B, multiple sequence alignment of the AA9 LPMOs investigated in this study. Positions involved in cello-oligosaccharide interactions and the His-brace motif are highlighted in green and blue, respectively. The positions of the primary and secondary coordination spheres of the copper are indicated with a blue or a magenta star, respectively. The L2, L3, L8, and LC loop regions are indicated with cyan, green, orange, and yellow bars below the sequences, respectively. LC, C-terminal; LPMOs, lytic polysaccharide monooxygenases.

Table 1

LsAA9A homologue sequences

Enzyme name	Organism	AA9 subcluster	Sequence identity to LsAA9A	Substitutions	
				Negative subsites	Positive subsites
LsAA9A	<i>Lentinus similis</i>	C38	-	n/a	n/a
PchAA9E	<i>Phanerochaete chrysosporium</i>	C38	76%	E148Q	
PcaAA9A	<i>Phanerochaete carmosa</i>	C38	74%	E148Q	
BaAA9A	<i>Bjerkandera adusta</i>	C38	69%	E148Q	N67D
AgAA9A	<i>Armillaria gallica</i>	C38	64%	D150Y, R159K	N67D
ScAA9A	<i>Schizophyllum commune</i>	C38	63%	Y203W	N67D
AoAA9A	<i>Aspergillus oryzae</i>	C29	55%	D150N, S151R, R159K	N67D
AfaA9C	<i>Aspergillus fumigatus</i>	C29	53%	D150N, S151R, R159K	N67D

Subcluster 38 (C38), subcluster 29 (C29), negative subsites (-4, -3, -2, and -1), positive subsites (+1 and +2).

For each AA9 enzyme name, the organism of origin and the cluster they belong to are given. The sequences are listed by their identity to LsAA9A and the substitutions affecting the subsite on either side of the scissile bond are indicated under *substitutions*.

subsites. ScAA9A has an Y203W substitution of the otherwise almost completely conserved platform Tyr involved in stacking interactions with the glycosyl unit in subsite -3. AgAA9A has a D150Y substitution, and because the Asp in LsAA9A interacts with the C6-hydroxyl of the glucosyl unit in subsite -3, it is possible that the Tyr in AgAA9A is able to interact with C6 substitutions on β -(1,4)-linked glucans such as the xylose decorations found in XG. Thus, both Y203W and D150Y likely affect substrate binding in subsite -3 (Fig. 1). AoAA9 and AfaA9 display some differences (D150N, S151R, or R159K) in the L8 loop, which is significantly shorter than the other six AA9s studied and similar to the equivalent loop in AA9 LPMOs

acting on insoluble cellulose (such as TaAA9A). Comparison of the TaAA9A, AoAA9A, and AfaA9C sequences indicate that an Arg residue is preserved at subsite -2, either at position 151 or 159.

To investigate the effect of natural variations at these functionally important positions, the catalytic domains of the seven selected AA9 LPMOs together with LsAA9A (control enzyme from the subcluster C38) were produced in *Pichia pastoris*. After purification, inductively coupled plasma mass spectrometry analyses indicated that the proteins were correctly Cu-loaded, in agreement with the H₂O₂ production rates observed using the Amplex red assay (Table S1).

Investigation of seven fungal AA9 LPMOs

Table 2
AA9 LPMO activities

Protein	Cellulose (PASC, Avicel)				XXXGXXXG oligosaccharide		Cell6	Subsites	Cell5	Subsites	Cell4	Subsites	Cell3	Subsites
	XG	GM	MLG	Xylan										
<i>LsAA9A</i>	++	++	++	++	(+)	++	++	-4 to +2	++	-3 to +2	+	-2 to +2	+	-1 to +2
<i>PchAA9E</i>	++	++	++	+	-	++	++	-4 to +2	++	-3 to +2	+	-2 to +2	-	-
<i>PcaAA9A</i>	++	++	++	+	-	++	++	-4 to +2	++	-3 to +2	+	-2 to +2	-	-
<i>BaAA9A</i>	++	-	-	-	-	+	++	-4 to +2	ND	-	-	-	ND	-
<i>AgAA9A</i>	++	++	+	+	-	++	++	-4 to +2	ND	-	(+)	(-2 to +2) (-3 to +1)	ND	-
<i>ScAA9A</i>	++	-	-	(+)	-	+	++	-3 to +3	ND	-	-	-	ND	-
<i>AoAA9A</i>	++	++	+	+	-	++	++	-4 to +2	++	-3 to +2	+	-2 to +2	ND	-
<i>AfAA9C</i>	++	++	+	+	-	++	++	-4 to +2	++	-3 to +2	+	-2 to +2	ND	-

XG, xyloglucan; GM, glucomannan; MLG, mixed-linked β -glucans; LPMO, lytic polysaccharide monoxygenase; cell4, cellotetraose; cell5, cellopentaose; cell6, cellohexaose; ND, not determined.

LsAA9A activities on cell5 and cell3 are inferred from literature (27, 28).

For each AA9 enzyme relative semiquantitative activities for one given substrate are indicated with “++”, “+”, “(+)”, or “-” for high activity, intermediate activity, little activity, or no activity, respectively. Subsites indicate which subsites the substrate occupy.

Table 3
Product profiles and initial rates of AA9 LPMO reactions with cello-oligosaccharides

Protein	Molar ratios after 2-h reaction with cell6 (%)			Change in Cell6 peak area (Δ area/min)	Fit of linear regression (R^2)	Rate of cell6 conversion ($\mu\text{M} \times \text{min}^{-1}$)	Rate of Cell6 conversion pr. enzyme (min^{-1})	Change in Cell5 peak area (Δ area/min)	Fit of linear regression (R^2)	Rate of cell5 conversion ($\mu\text{M} \times \text{min}^{-1}$)	Rate of Cell6 conversion pr. enzyme (min^{-1})
	Cell6	Cell4	Cell3								
<i>LsAA9A</i>	59	24	17	ND	ND	ND	ND	ND	-	-	-
<i>PchAA9E</i>	68	23	9	-0.12	0.94	3.0	0.8	-0.20	0.99	5.0	1.3
<i>PcaAA9A</i>	49	32	20	-0.14	0.98	3.5	0.9	-0.18	0.99	4.5	1.1
<i>BaAA9A</i>	83	11	6	ND	ND	ND	ND	ND	-	-	-
<i>AgAA9A</i>	68	20	12	-0.10	0.98	2.5	0.6	ND	-	-	-
<i>ScAA9A</i>	62	12	25	-0.13	0.97	3.3	0.8	ND	-	-	-
<i>AoAA9A</i>	57	31	12	-0.13	0.93	3.3	0.8	-0.18 ± 0.05	0.95 ± 0.03	4.5	1.1
<i>AfAA9C</i>	55	33	12	-0.15	0.95	3.8	0.9	-0.22 ± 0.02	0.97 ± 0.02	5.5	1.4

cell6, cellohexaose; cell5, cellopentaose; cell4, cellotetraose; LPMO, lytic polysaccharide monoxygenase; ND, not determined.

Areas of peaks corresponding to nonoxidized products and substrate (cell3, cell4 and cell6) after 2-h reaction were converted to molar ratios using the following relationship; $\text{area} = 4 \times [\text{cello-oligosaccharide}]$. The change in the substrate peak area (Δ cell5 or Δ cell6) were monitored using six time points; 0 min, 20 min, 40 min, 60 min, 90 min, 120 min, and linear regression was calculated based on these data points. The numerical value of the slopes was converted to molar rates, and the 100 \times dilution factor was applied to obtain the final rates of conversion of cell5 and cell6. The rate of cell6/cell5 conversion pr. enzyme is calculated by dividing the rate with the enzyme concentration (4 μM). Reactions with cell5 and *AoAA9A* or *AfAA9C* were run in triplicates. Rates were not calculated for *LsAA9A* and *BaAA9A* because the data points did not fit the regression properly.

The C29 and C38 AA9 LPMOs display activity on cellulose

The enzymatic activities of all *LsAA9A*-related LPMOs were first assayed on two cellulosic substrates, amorphous cellulose (phosphoric acid swollen cellulose) and crystalline cellulose (Avicel), with cysteine as a reductant because our previous studies suggest that the use of cysteine eases the interpretation of the chromatograms compared with, for example, ascorbate (10, 29). As expected, we found that all these enzymes displayed some activity on cellulose (Table 2 and Fig. S2). After only 4-h reactions, we detected nonoxidized cello-oligosaccharides (with a degree of polymerization [DP] of up to 6) and peaks corresponding to C4-oxidized products (with retention times of about 25 min and 40 min [29]), but no evidence of C1 oxidation (cell2^{ox}, cell3^{ox}, and cell4^{ox}). This clearly shows that all these AA9 LPMOs are C4-oxidizers when acting on substrates derived from cellulose. From overnight reactions with phosphoric acid swollen cellulose, we found that cellobiose (cell2), cellotriose (cell3), and cellotetraose (cell4) were the main products being formed, suggesting that these enzymes could be active on soluble cello-oligosaccharides (Fig. S2). The results obtained herein with *LsAA9A* produced in *P. pastoris* are thus in line with previous reports on the substrate specificity and regioselectivity of the same enzyme recombinantly produced in *A. oryzae* (27, 28). This indicates that the lack of methylation of His1 in

P. pastoris does not affect the product profile or the regioselectivity of the LPMO. Indeed, *LsAA9A* is a strict C4-oxidizer on cellulose and releases a range of short cello-oligosaccharides dominated by cell2 and cell3, in contrast to the longer products such as cell4, cellopentaose (cell5), and cell6 released by *TaAA9A*, which is not active on cello-oligosaccharides (27, 28).

The C29 and C38 AA9 LPMOs display activity on cello-oligosaccharides

We assayed the activity of the selected AA9 LPMOs on cello-oligosaccharides (cell3-cell6) in the presence of cysteine. All AA9 LPMOs tested were active on cell6 (Table 2), generating both cell3 and cell4 as the main nonoxidized products (Table 3 and Fig. 3), with constant rates over a 2-h reaction. We clearly found that no C1-oxidized products were formed and the chromatograms did indicate peaks corresponding to C4-oxidized species eluting around 25 and 40 min. Although the latter were not easily observable because of low sensitivity of species with late elution times, it strongly suggests that these AA9 LPMOs display a C4 regioselectivity in agreement with previous data on *LsAA9A*.

The majority of the AA9s tested produced cell4 as the major nonoxidized product followed by cell3, except for *ScAA9A*, which displayed a different profile because cell3 was the major

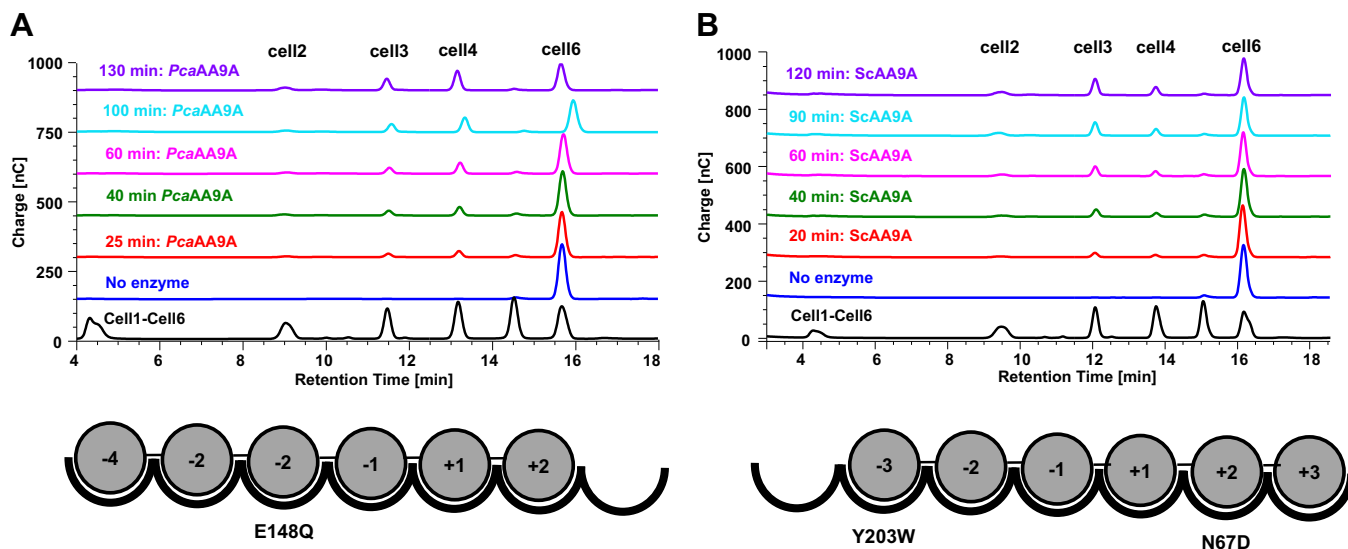


Figure 2. Time-course reaction of cellose degradation. Comparison of the preferred binding mode between the following: A, *Phanerochaete carnosae* AA9 (*PcaAA9A*) and B, *Schizophyllum commune* (*ScAA9A*). Schematic representations below the chromatograms indicate amino-acid substitution compared with *LsAA9A* and subsite organization.

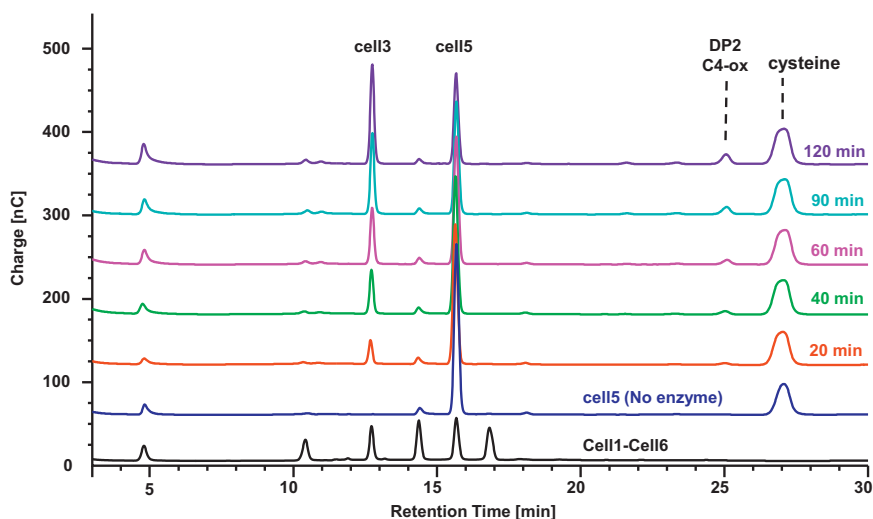


Figure 3. Time-course reaction of cellopentaose conversion exemplified by *PchAA9E*. Reactions with cellopentaose (cell5) analyzed after 20, 40, 60, 90, and 120 min. Cleavage of cell5 by *PchAA9E* produced cell3 (ca. 13-min retention time) and C4-oxidized cellobiose (ca. 25 min retention time) indicating that these enzymes are C4 oxidizers binding cell5 in a single binding mode. cell3, cellobiose.

product (Fig. 2, Tables 2 and 3). This interesting observation indicates that the majority of the AA9 enzymes can bind cell6 preferably from subsite -4 to $+2$ in agreement with previous biochemical and structural reporting on *LsAA9A* (e.g. in supplementary figures 1 and 4 of [27]). However, based on our observations, we believe that the preferred binding mode of *ScAA9A* is from subsite -3 to $+3$. The rates of substrate conversion (and product formation although not shown) were determined and under our experimental conditions, we found that the majority of AA9 LPMOs converted cell6 with relatively similar constant rates of around 1 min^{-1} (Table 3).

When investigating LPMO activity on cellooligosaccharides smaller than cell6 ($<DP6$), we found, just like for *LsAA9A*, that *PchAA9E*, *PcaAA9A*, *AoAA9A*, and *AfAA9C* were clearly able to degrade cell5 and cell4, whereas

weak activity was observed for *AgAA9A*. *PchAA9E* and *PcaAA9A*, the AA9 LPMOs most closely related to *LsAA9A*, showed no activity with cell3 as substrate unlike *LsAA9A* (27). With cell5 as substrate, we found that *PchAA9E*, *PcaAA9A*, *AoAA9A*, and *AfAA9C* (exemplified by *PchAA9E*, Fig. 3) generated cell3 as the major nonoxidized product and C4-oxidized DP2 products (and also a minor peak corresponding to glucose emerged during 2-h reaction). This indicates that the preferred binding mode of cell5 is from subsites -3 to $+2$ similarly to *LsAA9A*. We monitored the conversion of cell5 and the corresponding formation of cell3, which occurred at a constant rate over a 2-h time course (Table 3 and Fig. 3). Again, we found that the four enzymes appeared to convert the cell5 substrate at similar rates of around 1 min^{-1} (Table 3). The small formation of glucose could arise from some scarce

Investigation of seven fungal AA9 LPMOs

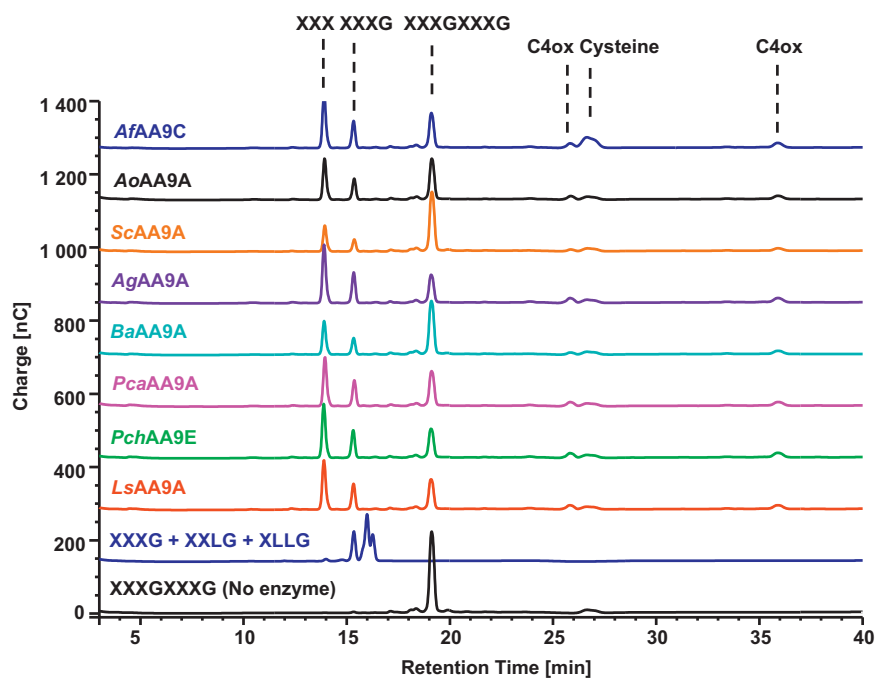


Figure 4. Activity of AA9 LPMOs on xyloglucan. Cleavage of XXXGXXXG by the AA9 LPMOs after a 24-h reaction. The major nonoxidized product is XXX, but also XXXG is formed indicating that an unsubstituted glucosyl unit (G) is preferred in subsite +1 and that a C6-xylose substituted glucosyl unit (X) is tolerated. The XG-oligosaccharide standards were previously found to elute in following order—XXXG, XXLG, and XLLG (24, 30). LPMOs, lytic polysaccharide monoxygenases; XG, xyloglucan.

cell3 degradation that occurred under these experimental conditions, although we did not observe activity when running experiments with only cell3 as a substrate.

With a cell4 substrate, we found that the four enzymes produced mainly cell2, confirming the binding mode from the -2 to $+2$ subsites, as previously reported for *LsAA9A* (27, 28). In contrast, *AgAA9A* appeared to equally produce both cell2 and glucose, although the activity was low, indicating binding from both -2 to $+2$ and -3 to $+1$ (Table 2). Incubating any of the substrates in the absence of enzyme or replacing the enzyme with equimolar Cu-acetate resulted in no substrate conversion or product formation.

The C29 and C38 AA9 LPMOs display activity on hemicellulose-derived substrates

Next, we tested the ability of the enzymes to degrade an XG polysaccharide from tamarind seeds. The reaction products were analyzed with chromatography using XXXG, XXLG, and XLLG oligosaccharides as standards (nomenclature after (41) where G and X denote β -(1,4)-linked glucosyl units either nonsubstituted or with xylosyl substitutions at the C6 position, respectively). For the majority of AA9s, we observed significant activity except for *BaAA9A* and *ScAA9A* where activity could not be detected even after overnight incubation (Fig. S4A). Among the products from 1-h reactions (Fig. S4B), three peaks with retention times around 15 min could correspond to short nonoxidized oligosaccharides (such as XXXG) and peaks with retention times 20 to 25 min, which could correspond to longer native or C4-oxidized oligosaccharides (after comparison with [24]). After overnight reactions (ca. 22 h, Fig. S4A), these peaks were significantly

smaller, indicating a possible activity of these enzymes on XG oligosaccharides. Therefore, we tested the ability of these AA9 LPMOs to degrade an XG XXXGXXXG tetradecasaccharide. Reactions with XXXGXXXG showed that all AA9 (including *BaAA9A* and *ScAA9A*) cleaved XXXGXXXG, which lead to a small peak corresponding to XXXG and a larger peak corresponding likely to an XXX hexasaccharide (Fig. 4). This suggests that these AA9 LPMOs preferentially bind the XXXGXXXG nonreducing end in subsite -3 and with a nonsubstituted glucosyl unit in the $+1$ subsite, which would generate XXX and C4-oxidized GXXXG species, as also reported by Agger *et al.* (24). They can also accommodate a xylose-substituted glucosyl unit in subsite $+1$ and bind the XXXGXXXG nonreducing end in subsite -4 producing a nonoxidized and a C4-oxidized XXXG species. We also speculated that the D150Y substitution near subsite -3 in *AgAA9A* (Fig. 5) could enhance binding of a C6-substituted glucosyl unit, and thus, *AgAA9A* would have the strongest preference for binding XXXGXXXG. Reactions with XXXGXXXG and *AgAA9A* or *LsAA9A* were run in parallel for 20 min, 2 h, and 24 h (Fig S5); however, the results of the time-course reaction with XXXGXXXG did not reveal any striking difference between *AgAA9A* and *LsAA9A*.

Finally, we tested the ability of the AA9 enzymes to cleave other hemicellulose substrates such as MLG, GM, and xylan. For many of the AA9s, products following enzymatic cleavage were detectable from reactions with MLG and GM. However, for *ScAA9A*, we only found weak activity on MLG and for *BaAA9A*, no clear activity on either of MLG or GM (Table 2). No clear activity on xylan or xylohexaose was observed for any of the *LsAA9A*-related LPMOs.

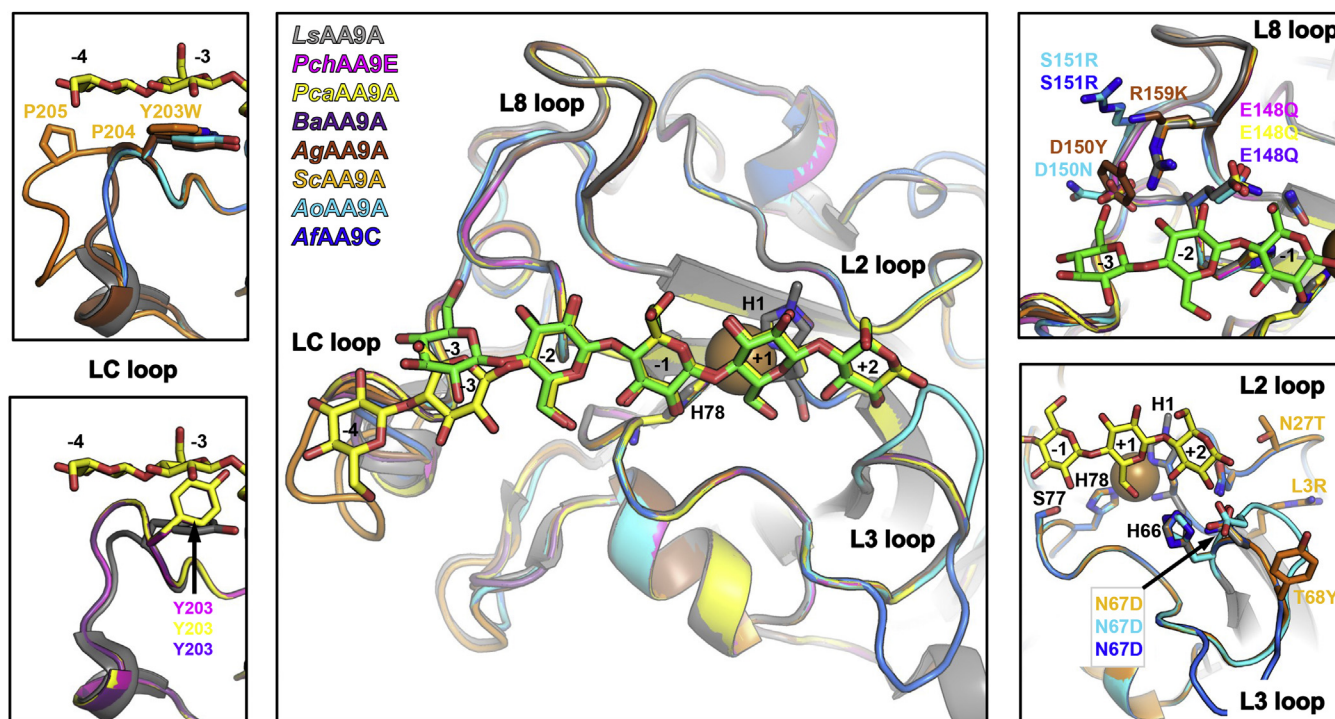


Figure 5. Homology models of AA9 LPMOs. Loop elements of each model are colored as follows—*PchAA9E* (magenta), *PcaAA9A* (yellow), *BaAA9A* (purple), *AgAA9A* (brown), *ScAA9A* (orange), *AoAA9A* (cyan), and *AfAA9C* (blue). *LsAA9A* structures are shown transparent in gray. All β -strands are in gray, and the L2, L3, L8, and LC loops are indicated with black labels. The insets highlight differences of the models in loops L2 and L3 (lower right panel), L8 (upper right panel), and LC (left panels). Labels are colored with according to the color of the model. Black labels apply to *LsAA9A*. Cellopentaose (from the *LsAA9A*-cell5 complex PDB 5NLS) and cellohexaose (from the *LsAA9A*-cell6 complex PDB 5dsACI) are shown in green and yellow, respectively. LC, C-terminal; LPMOs, lytic polysaccharide monoxygenases.

Homology models of the studied LPMOs

To better interpret and discuss the biochemical results, homology modeling was carried out using *LsAA9A* as the template. All models were highly reliable and with reasonable geometric quality (Table S2) according to Swiss-Model scores (42). Although still reliable, the *AoAA9A* model had the lowest quality as expected, given its lowest sequence identity (~55 %) to the template (Table 1 and Fig. S6A). The LC loops bearing the Tyr/Trp residue expected to interact with the substrate (see Discussion) are the regions of the homology models that are structurally most different from the *LsAA9A* template. However, some caution must be applied in interpretation because these regions are also less reliably modeled (Fig S6).

Discussion

Here, we have investigated substrate specificities of seven fungal AA9 LPMOs from two related phylogenetic subclusters (C29 and C38) and shown activity on both cello-oligosaccharide- and hemicellulose-derived substrates. Our results support the notion that activity on short cello-oligosaccharides correlate with the activity on plant cell wall β -(1,4)-linked hexose chains. All of the LPMOs behaved relatively similarly to the canonical *LsAA9A* in terms of regioselectivity, ability to degrade cello-oligosaccharides and hemicelluloses containing β -(1,4)-linked D-glucose in contrast to AA9 from the other groups, which are mostly only active on cellulose. Our results show that all of the investigated LPMOs

could bind cell6 from both subsite -4 to +2 and -3 to +3, indicating that the LPMOs in fact recognize five β -(1,4)-linked glucosyl units as in cell5. Indeed, we found a single binding mode for cell5 (from subsites -3 to +2), and both cell5 and cell6 are degraded with similar rates (indicating that subsites -4 and +3 are not strong binding sites). The very similar and low rates measured could be an indication of H_2O_2 being a limiting factor. It would be interesting to further investigate the activity of these LPMOs using H_2O_2 as cosubstrate under more controlled conditions. Based on the set of AA9 LPMOs investigated in this study, we conclude that in general, an extended L3 and an L8 loop both with polar/charged amino acid residues are determinants of cello-oligosaccharide activity in AA9 LPMOs. These features are present in AA9 LPMOs from C29 and C38 and small sequence differences within the L3, L8, and LC loops do not significantly impact the activity.

Although the AA9 LPMOs investigated here are overall similar, in-depth biochemical analyses revealed some subtle differences between the enzymes studied in terms of activity and substrate binding. For instance, *LsAA9A* is able to cleave cell3 (bound by Glu148 and Asn67 in subsites -1 to +2, respectively), unlike the other AA9 LPMOs investigated here. We found that *PchAA9E* and *PcaAA9A* (most closely related to *LsAA9A*) cleave cell4 as the smallest oligosaccharide substrate (from subsites -2 to +2). Structural modeling shows that Gln148 of *PchAA9E* and *PcaAA9A* align well with the *LsAA9A* Glu148. It is possible that E148Q alter the H-bond, which weakens cell3 binding, which could explain the absence of cell3 activity (Fig. 5).

Investigation of seven fungal AA9 LPMOs

It also appears that N67D and R159K lead to loss of cell4 activity unless compensated by other substitutions in the L8 loop such as D150N and S151R found in *AoAA9A* and *AfAA9C* (Fig. 5 and Fig. S6A). Our results also suggest that the double substitution N67D/E148Q negatively affects cello-oligosaccharide binding because the *BaAA9A* enzyme only showed activity on cell6 and not on cello-oligosaccharides < DP6 (Table 2).

The *ScAA9A* enzyme displayed a different product profile by binding cell6 from -3 to +3. A weakened -4 subsite or a strengthened +3 subsite could explain this observation. Indeed, *ScAA9A* displays a Trp in the LC loop instead of the otherwise conserved substrate-interacting platform Tyr. Based on the *ScAA9A* model, these aromatic side chains likely make a similar interaction with cello-oligosaccharides, although two Pro residues (Pro204 and Pro205, not found in the other investigated enzymes) appear to affect the conformation of the LC loop (Fig. 1B, Fig. 5, and Fig. S6C). We believe that these subtle differences could alter the accommodation of the substrates in the -4 subsite and that the Trp preferably interacts with the terminal glucosyl unit at the nonreducing end in subsite -3. The *ScAA9A* 3D model also indicate that the T68Y and N27T substitutions (in loops L3 and L2, respectively), which are the neighboring residues of those making up subsite +2, and the L3R could play a role in binding of the terminal glucosyl unit at the reducing end in subsite +3 (Fig. 5, bottom right inset). Both of these explanations would lead to preferential binding of cell6 from -3 to +3.

A recently determined structure of *CvAA9A* (which shares 45% sequence identity with *LsAA9A* and also belongs to subcluster C38) with cell6 bound between subsite -3 and +3 (PDB code 6YDE) shows that the *CvAA9A* Arg3 is involved in a water-mediated hydrogen-bonding network in contact with the glucosyl unit at subsite +3 (33). In addition, both biochemical and structural analyses show that *CvAA9A* binds cell4 from -3 to +1 (PDB code 6YDC) unlike the AA9 LPMOs investigated here.

We also found some differences when investigating the conversion of hemicellulose substrates. In the *AgAA9A* 3D model, a Tyr (D150Y) is positioned next to the Tyr platform near subsite -3 (Fig. 5 and Fig. S6B) and could play a role in XG binding through interaction with xylose substitutions at the C6-hydroxyl of the glucosyl unit. However, the XG activity of *AgAA9A* was not markedly higher than that of other AA9 LPMOs characterized here (Fig. S4). Somewhat puzzling, however, we found that *BaAA9* and *ScAA9A* cleaved an XXXGXXXG oligosaccharide but did not show any activity on the XG polysaccharide substrate, unlike the rest of the characterized AA9 LPMOs. Finally, although *LsAA9A* is able to cleave xylan and xylohexaose (albeit much less efficient than cellulose substrates), we did not find any significant activity on these substrates with any of the other C29 or C38 AA9s investigated here. Thus, *LsAA9A* remains the only AA9 LPMO with reported activity on isolated xylan (as found here and previously [28]). The AA9 LPMOs from subclusters C29 and C38 (modular AA9-CBM1 proteins or single-domain AA9 enzymes, respectively) characterized in this study all originate from fungal species commonly associated with saprotrophic

and/or plant parasitic lifestyles. Many of these are *Agaricomycetes* fungal species that display multiple AA9 genes, but on average only one or two putative cello-oligosaccharide-active AA9 LPMOs. During this study, we have also noted that some AA9 sequences, grouped in the neighboring subcluster C27, have similar putative cello-oligosaccharide binding features and are appended to a module of unknown function. These sequences originate mostly from plant pathogen species of the *Dothideomycetes* class, and it would be interesting to see if there is a correlation between cello-oligosaccharide specificity and pathogenicity.

We and others have previously observed a correlation between LPMO activity on hemicelluloses containing β -(1,4)-linked D-glucose (24, 25, 29, 35) and cello-oligosaccharides for some AA9s (e.g. *NcAA9C*, *PaAA9H*, and *CtPMO1*) from other subclusters (C21 and C23, which contain many *Sordariomycetes* species and are not closely related to C38 or C29) (Fig. S1). These AA9 LPMOs behave similarly to the AA9 LPMOs investigated in this study. This shows that the LPMO activity on both cello-oligosaccharides and hemicelluloses is not limited to AA9s in a few closely related phylogenetic subclusters but is found more widely in a number of fungal phylogenetic classes in species with different lifestyles. Interestingly, a recent transcriptomic study on *P. carnososa* showed that the gene encoding *PcaAA9A* (studied herein) was expressed during several days on growth on white spruce (*Picea glauca*) and aspen (*Populus tremuloides*) (43). In another study on *Aspergillus nidulans*, the gene encoding AN1602 (44) (an AA9-CBM1 protein displaying 65% sequence identity with *AoAA9* and *AfAA9*) was highly transcribed during growth on cellulose and xylan, and the enzyme was shown to be active on both cell6 and XG (44, 45). Recently, XG and cell6 activities have also been reported for an AA9-CBM1 LPMO from *Aspergillus tamaritii* (*AtAA9A*) for which the catalytic AA9 domain shares about 80% sequence identity with *AoAA9A* and *AfAA9C* (46).

It seems that many wood-decaying fungal species use an arsenal of AA9 LPMOs with at least one AA9 member that is able to recognize a short β -(1,4)-linked glucan motif conferring activity on cello-oligosaccharides and hemicellulosic substrates. The functional features of the fungal AA9 LPMOs studied herein do not fit with the common belief that LPMOs are specifically targeting crystalline regions of cellulose fibrils. Rather, C29 and C38 LPMOs seem to target a variety of complex motifs within the plant cell wall. Whatever is their biological function, we have shown that it is possible, from their sequence, to predict their LPMO activity toward cello-oligosaccharides and hemicellulose substrates, although their precise substrate specificity remains difficult to predict. In addition, the AA9 LPMOs displaying a single oligosaccharide-binding mode could be useful model systems for further mechanistic investigations.

Experimental procedures

Cloning and expression of AA9 LPMOs

All proteins were produced using the in-house 3PE Platform (*Pichia Pastoris* Protein Express; www.platform3pe.com/) (47). Genes encoding *LsAA9A* (GenBank ALN96977.1), *AfAA9C*

(EAL85444.1), AoAA9A (UniProt Q2US83), PchAA9E (JGI protein ID 2934397), PcaAA9A (261285), BaAA9A (353490), AgAA9A (500811), and ScAA9A (2617723) were synthesized after codon optimization for *P. pastoris* (GeneScript, Piscataway, NJ) and inserted into the vector pPICZαA (Invitrogen, Cergy-Pontoise, France) using BstBI and XbaI restriction sites in frame with the C-terminal (His)₆-tag.

P. pastoris strain X33 and the pPICZαA vector are components of the *P. pastoris* Easy Select Expression System (Invitrogen). The resulting pPICZαA plasmid containing the AA9 gene was linearized with PmeI, and competent *P. pastoris* X33 was transformed using electroporation as described elsewhere ([29, 47] and reference herein). Zeocin-resistant *P. pastoris* transformants were then screened for protein production (47). Protein candidates suitable for upscaled production were produced in 2-L flasks in which cells were grown in 500-ml (buffered complex glycerol medium containing 1 ml L⁻¹ Pichia trace elements (PTM₄) salts and buffered with 100-mM phosphate, pH 6.0) at 30 °C in an orbital shaker (200 rpm) for 16 h until an absorbance of ~ 2 to 6 at 600 nm. The cells were pelleted by centrifugation (10 min at 2700g) at room temperature and resuspended in 100 ml of (buffered complex methanol medium) containing 1 ml L⁻¹ (PTM₄) salts at 20 °C in an orbital shaker (200 rpm) for 3 days with daily addition of 3% methanol (v/v). The culture supernatants were collected by centrifugation at 2700g for 10 min at 4 °C. The pH was adjusted to 7.8 just before purification and filtered using 0.22-μm filters (Millipore, Molsheim, France).

Purification and copper loading of AA9 LPMOs

Purification was performed using an AKTExpress system (GE healthcare). The supernatants were loaded onto 5-ml His Trap HP columns (GE healthcare, Buc, France) equilibrated in 50-mM Tris-HCl, pH 7.8, 150-mM NaCl, and 10-mM imidazole (buffer A), and sample loading was finished by an additional two column volumes of buffer A. The proteins were eluted with imidazole in a stepwise manner with 2% (10 mM), 50% (250 mM), and 100% (500 mM) of buffer B (50-mM Tris HCl, pH 7.8, 150-mM NaCl, 500-mM imidazole). Fractions containing recombinant enzymes were pooled and concentrated with a 10-kD vivaspin concentrator (Sartorius, Palaiseau, France) to remove imidazole and buffer exchanged in 50-mM 2-(N-morpholino)ethanesulfonic acid buffer, pH 6.0. The concentration of purified proteins was determined by absorption at 280 nm using a Nanodrop ND-2000 spectrophotometer (Thermo Fisher Scientific), with calculated molecular mass and molar extinction coefficients derived from the sequences. Proteins were loaded onto 10% SDS-PAGE gels (Thermo Fisher Scientific, IL), which were stained with Coomassie Blue. The molecular mass under denaturing conditions was determined with reference standard proteins (PageRuler Prestained Protein Ladder, Thermo Fisher Scientific). The concentrated proteins were incubated with Cu-acetate (slight in excess) for either 1 h at room temperature or overnight at 4 °C. Excess of copper was removed by loading onto a HiLoad 16/600 Superdex 75 Prep Grade column (GE healthcare) and separated in 20-mM Na-acetate, pH 5.5, and

100-mM NaCl. The copper-loaded and purified batches (prepared in 20 mM Na-acetate pH 5.5 and 100 mM NaCl) were used for the Amplex Red Assay (48, 49), inductively coupled plasma mass spectrometry analysis (as previously described [13]), and enzymatic reactions.

AA9 LPMO enzymatic reactions

Reactions with AA9 enzymes (at 4-μM concentration) were carried out at 50 °C at 850 rpm in 20-mM Na-acetate, pH 5.5, 100-mM NaCl in the presence of 1 mM cysteine, chosen in preference to other reducing agents for easier interpretation of the chromatograms. For polysaccharides, incubation times were either 1 h or overnight (approximately 20 h) and reaction mixtures were diluted ten times before injection. XG from tamarind seeds (CAS No. 37294-28-3) was purchased from Megazyme, and XXXGXXXG was kindly provided by H. Brumer (UBC, Vancouver, Canada). It was prepared as described in (50). For cello-oligosaccharides (initial concentration of 1 mM), time-course reactions were carried out with time points at 20, 40, 60, 90, and 120 min, and the reaction mixtures injected to the system were x100 dilutions. For the C29 AA9s, the 2 h time-course reactions with cell5 were run in triplicates. For time-course experiments with cell5, the peak areas of the different reaction time points were plotted against time and the slope was converted to molar rates. Control reactions with LPMO and a cello-oligosaccharide in the absence of an electron donor showed no product formation. For all substrates, negative controls were run both without enzyme and in the presence of Cu-acetate in equimolar concentrations to the enzymes (*i.e.* 4 μM). None of the negative controls with Cu-acetate produced peaks above the background.

Analysis of carbohydrate products

Analysis of carbohydrate products (both oxidized and nonoxidized) after incubation of polysaccharide and oligosaccharide substrates with AA9 LPMOs was performed using high-performance anion-exchange chromatography coupled with pulsed amperometric detection as described in (51). By the use of standards (Megazyme), peak areas were converted to molar concentration. A peak corresponding to the electron donating redox partner cysteine was observed with retention time of 26 to 27 min. Peaks at retention times corresponding to oxidized products have been previously reported (29).

Homology modeling

Swiss-Model (<https://swissmodel.expasy.org/>) (42) was chosen because we found this method to better handle the methylated His and the position of the Tyr platform (Tyr203 in LsAA9A) than other available methods (*e.g.* the Phyre2 server (<http://www.sbg.bio.ic.ac.uk/>) [52]). Swiss-Model runs through the server at <https://swissmodel.expasy.org/> with template PDB 5ACH (the closest available for all investigated enzymes) (42). GQME and QMEAN Z-scores used to assess model quality are summarized in (Table S2). The lowest scores in terms of reliability and geometry were GQME of 0.77 and QMEAN Z-score of -2.09, which are still very reasonable for a

Investigation of seven fungal AA9 LPMOs

useful model. MolProbity overall scores are similar for models and template, with models generally having slightly worse Ramachandran statistics.

Data availability

All data generated or analyzed during this study are contained within this article.

Acknowledgments—The authors thank Florence Chaspoul from IMBE (Institut Méditerranéen de Biodiversité et d'Ecologie, CNRS Marseille) for inductively coupled plasma mass spectrometry analysis and Harry Brumer for providing the XXXGXXXG substrate.

Author contributions—K. E. H. F. and J. G. B. designed the study. K. E. H. F. carried out most of the experimental work. K. E. H. F. and B. H. carried out the bioinformatics analysis. K. E. H. F. and M. H. expressed and purified recombinant lytic polysaccharide mono-oxygenases. K. E. H. F. carried out enzyme assays. K. E. H. F. and S. G. analyzed enzymatic products with high-performance anion-exchange chromatography coupled with pulsed amperometric detection. L. L. L. made homology models. K. E. H. F. and J. G. B. wrote the manuscript with contribution from the coauthors. The manuscript was approved by all coauthors before submission.

Funding and additional information—This work was supported by Carlsberg Foundation Internationalisation Postdoc Fellowship Grants CF16-0673 and CF17-0533 (to K. E. H. F.), EU Marie-Curie FP7 COFUND People Programme (grant agreement 609398), through an AgreeSkills Fellowship (to K. E. H. F.), and Novo Nordisk Foundation Grant NNF17SA0027704 (L. L. L.).

Conflict of interest—The authors declare that they have no conflicts of interest with the contents of this article.

Abbreviations—The abbreviations used are: CBM, carbohydrate-binding module; Cell1, glucose; Cell2, cellobiose; Cell3, cellotriose; Cell4, cellotetraose; Cell5, cellopentaose; Cell6, cellohexaose; DP, degree of polymerization; GM, glucomannan; His-brace, Histidine brace; ICP-MS, inductively coupled plasma mass spectrometry; LC, C-terminal; LPMO, lytic polysaccharide mono-oxygenase; MLGs, mixed-linkage β -glucans; XG, xyloglucan.

References

1. Johansen, K. S. (2016) Lytic polysaccharide mono-oxygenases: the microbial power tool for lignocellulose degradation. *Trends Plant. Sci.* **21**, 926–936
2. Tandrup, T., Frandsen, K. E. H., Johansen, K. S., Berrin, J.-G., and Lo Leggio, L. (2018) Recent insights into lytic polysaccharide mono-oxygenases (LPMOs). *Biochem. Soc. Trans.* **46**, 1431–1447
3. Vaaje-Kolstad, G., Westereng, B., Horn, S. J., Liu, Z., Zhai, H., Sorlie, M., and Eijsink, V. G. (2010) An oxidative enzyme boosting the enzymatic conversion of recalcitrant polysaccharides. *Science* **330**, 219–222
4. Bissaro, B., Rohr, A. K., Muller, G., Chylenski, P., Skaugen, M., Forsberg, Z., Horn, S. J., Vaaje-Kolstad, G., and Eijsink, V. G. H. (2017) Oxidative cleavage of polysaccharides by monocopper enzymes depends on H₂O₂. *Nat. Chem. Biol.* **13**, 1123–1128
5. Walton, P. H., and Davies, G. J. (2016) On the catalytic mechanisms of lytic polysaccharide mono-oxygenases. *Curr. Opin. Chem. Biol.* **31**, 195–207
6. Lenfant, N., Hainaut, M., Terrapon, N., Drula, E., Lombard, V., and Henrissat, B. (2017) A bioinformatics analysis of 3400 lytic polysaccharide oxidases from family AA9. *Carbohydr. Res.* **448**, 166–174
7. Levasseur, A., Drula, E., Lombard, V., Coutinho, P. M., and Henrissat, B. (2013) Expansion of the enzymatic repertoire of the CAZY database to integrate auxiliary redox enzymes. *Biotechnol. Biofuels* **6**, 41
8. Crouch, L. I., Labourel, A., Walton, P. H., Davies, G. J., and Gilbert, H. J. (2016) The contribution of non-catalytic carbohydrate binding modules to the activity of lytic polysaccharide mono-oxygenases. *J. Biol. Chem.* **291**, 7439–7449
9. Courtade, G., Forsberg, Z., Heggset, E. B., Eijsink, V. G. H., and Aachmann, F. L. (2018) The carbohydrate-binding module and linker of a modular lytic polysaccharide mono-oxygenase promote localized cellulose oxidation. *J. Biol. Chem.* **293**, 13006–13015
10. Chalak, A., Villares, A., Moreau, C., Haon, M., Grisel, S., d'Orlando, A., Herpoël-Gimbert, I., Labourel, A., Cathala, B., and Berrin, J.-G. (2019) Influence of the carbohydrate-binding module on the activity of a fungal AA9 lytic polysaccharide mono-oxygenase on cellulosic substrates. *Biotechnol. Biofuels* **12**, 206
11. Lombard, V., Golaconda Ramulu, H., Drula, E., Coutinho, P. M., and Henrissat, B. (2014) The carbohydrate-active enzymes database (CAZY) in 2013. *Nucleic Acids Res.* **42**, D490–D495
12. Filiatrault-Chastel, C., Navarro, D., Haon, M., Grisel, S., Herpoël-Gimbert, I., Chevret, D., Fanuel, M., Henrissat, B., Heiss-Blanquet, S., Margeot, A., and Berrin, J. G. (2019) AA16, a new lytic polysaccharide mono-oxygenase family identified in fungal secretomes. *Biotechnol. Biofuels* **12**, 55
13. Couturier, M., Ladeveze, S., Sulzenbacher, G., Ciano, L., Fanuel, M., Moreau, C., Villares, A., Cathala, B., Chaspoul, F., Frandsen, K. E., Labourel, A., Herpoël-Gimbert, I., Grisel, S., Haon, M., Lenfant, N., *et al.* (2018) Lytic xylan oxidases from wood-decay fungi unlock biomass degradation. *Nat. Chem. Biol.* **14**, 306–310
14. Hemsworth, G. R., Henrissat, B., Davies, G. J., and Walton, P. H. (2014) Discovery and characterization of a new family of lytic polysaccharide mono-oxygenases. *Nat. Chem. Biol.* **10**, 122–126
15. Lo Leggio, L., Simmons, T. J., Poulsen, J.-C. N., Frandsen, K. E. H., Hemsworth, G. R., Stringer, M. A., von Freiesleben, P., Tovborg, M., Johansen, K. S., De Maria, L., Harris, P. V., Soong, C.-L., Dupree, P., Tryfona, T., Lenfant, N., *et al.* (2015) Structure and boosting activity of a starch-degrading lytic polysaccharide mono-oxygenase. *Nat. Commun.* **6**, 5961
16. Vu, V. V., Beeson, W. T., Span, E. A., Farquhar, E. R., and Marletta, M. A. (2014) A family of starch-active polysaccharide mono-oxygenases. *Proc. Natl. Acad. Sci. U. S. A.* **111**, 13822–13827
17. Harris, P. V., Welner, D., McFarland, K. C., Re, E., Navarro Poulsen, J. C., Brown, K., Salbo, R., Ding, H., Vlasenko, E., Merino, S., Xu, F., Cherry, J., Larsen, S., and Lo Leggio, L. (2010) Stimulation of lignocellulosic biomass hydrolysis by proteins of glycoside hydrolase family 61: structure and function of a large, enigmatic family. *Biochemistry* **49**, 3305–3316
18. Sabbadin, F., Hemsworth, G. R., Ciano, L., Henrissat, B., Dupree, P., Tryfona, T., Marques, R. D. S., Sweeney, S. T., Besser, K., Elias, L., Pesante, G., Li, Y., Dowle, A. A., Bates, R., Gomez, L. D., *et al.* (2018) An ancient family of lytic polysaccharide mono-oxygenases with roles in arthropod development and biomass digestion. *Nat. Commun.* **9**, 756
19. Yadav, S. K., Archana, Singh, R., Singh, P. K., and Vasudev, P. G. (2019) Insecticidal fern protein Tma12 is possibly a lytic polysaccharide mono-oxygenase. *Planta* **249**, 1987–1996
20. Quinlan, R. J., Sweeney, M. D., Lo Leggio, L., Otten, H., Poulsen, J. C. N., Johansen, K. S., Krogh, K. B. R. M., Jorgensen, C. I., Tovborg, M., Anthonsen, A., Tryfona, T., Walter, C. P., Dupree, P., Xu, F., Davies, G. J., *et al.* (2011) Insights into the oxidative degradation of cellulose by a copper metalloenzyme that exploits biomass components. *Proc. Natl. Acad. Sci. U. S. A.* **108**, 15079–15084
21. Frandsen, K. E. H., and Lo Leggio, L. (2016) Lytic polysaccharide mono-oxygenases: a crystallographer's view on a new class of biomass-degrading enzymes. *IUCr* **3**, 448–467
22. Frandsen, K. E. H., Tovborg, M., Jorgensen, C. I., Spodsberg, N., Rosso, M. N., Hemsworth, G. R., Garman, E. F., Grime, G. W., Poulsen, J. C. N., Batth, T. S., Miyauchi, S., Lipzen, A., Daum, C., Grigoriev, I. V., Johansen, K. S., *et al.* (2019) Insights into an unusual Auxiliary Activity 9 family member lacking the histidine brace motif of lytic polysaccharide mono-oxygenases. *J. Biol. Chem.* **294**, 17117–17130

23. Vaaje-Kolstad, G., Forsberg, Z., Loose, J. S. M., Bissaro, B., and Eijsink, V. G. H. (2017) Structural diversity of lytic polysaccharide monoxygenases. *Curr. Opin. Struct. Biol.* **44**, 67–76
24. Agger, J. W., Isaksen, T., Varnai, A., Vidal-Melgosa, S., Willats, W. G., Ludwig, R., Horn, S. J., Eijsink, V. G., and Westereng, B. (2014) Discovery of LPMO activity on hemicelluloses shows the importance of oxidative processes in plant cell wall degradation. *Proc. Natl. Acad. Sci. U. S. A.* **111**, 6287–6292
25. Isaksen, T., Westereng, B., Aachmann, F. L., Agger, J. W., Kracher, D., Kittl, R., Ludwig, R., Haltrich, D., Eijsink, V. G., and Horn, S. J. (2014) A C4-oxidizing lytic polysaccharide monoxygenase cleaving both cellulose and cello-oligosaccharides. *J. Biol. Chem.* **289**, 2632–2642
26. Huttner, S., Varnai, A., Petrovic, D. M., Bach, C. X., Kim Anh, D. T., Thanh, V. N., Eijsink, V. G. H., Larsbrink, J., and Olsson, L. (2019) Specific xylan activity revealed for AA9 lytic polysaccharide monoxygenases of the thermophilic fungus *Malbranchea cinnamomea* by functional characterization. *Appl. Environ. Microbiol.* **85**, 1408–1419
27. Frandsen, K. E., Simmons, T. J., Dupree, P., Poulsen, J. C., Hemsworth, G. R., Ciano, L., Johnston, E. M., Tovborg, M., Johansen, K. S., von Freileben, P., Marmuse, L., Fort, S., Cottaz, S., Driguez, H., Henrissat, B., et al. (2016) The molecular basis of polysaccharide cleavage by lytic polysaccharide monoxygenases. *Nat. Chem. Biol.* **12**, 298–303
28. Simmons, T. J., Frandsen, K. E. H., Ciano, L., Tryfona, T., Lenfant, N., Poulsen, J. C., Wilson, L. F. L., Tandrup, T., Tovborg, M., Schnorr, K., Johansen, K. S., Henrissat, B., Walton, P. H., Lo Leggio, L., and Dupree, P. (2017) Structural and electronic determinants of lytic polysaccharide monoxygenase reactivity on polysaccharide substrates. *Nat. Commun.* **8**, 1064
29. Bennati-Granier, C., Garajova, S., Champion, C., Grisel, S., Haon, M., Zhou, S., Fanuel, M., Ropartz, D., Rogniaux, H., Gimbert, I., Record, E., and Berrin, J. G. (2015) Substrate specificity and regioselectivity of fungal AA9 lytic polysaccharide monoxygenases secreted by *Podospora anserina*. *Biotechnol. Biofuels* **8**, 90
30. Fanuel, M., Garajova, S., Ropartz, D., McGregor, N., Brumer, H., Rogniaux, H., and Berrin, J.-G. (2017) The *Podospora anserina* lytic polysaccharide monoxygenase PaLPMO9H catalyzes oxidative cleavage of diverse plant cell wall matrix glycans. *Biotechnol. Biofuels* **10**, 63
31. Basotra, N., Dhiman, S. S., Agrawal, D., Sanj, R. K., Tsang, A., and Chadha, B. S. (2019) Characterization of a novel lytic polysaccharide monoxygenase from *Malbranchea cinnamomea* exhibiting dual catalytic behavior. *Carbohydr. Res.* **478**, 46–53
32. Borisova, A. S., Isaksen, T., Dimarogona, M., Kognole, A. A., Mathiesen, G., Varnai, A., Rohr, A. K., Payne, C. M., Sorlie, M., Sandgren, M., and Eijsink, V. G. (2015) Structural and functional characterization of a lytic polysaccharide monoxygenase with broad substrate specificity. *J. Biol. Chem.* **290**, 22955–22969
33. Tandrup, T., Tryfona, T., Frandsen, K. E. H., Johansen, K. S., Dupree, P., and Lo Leggio, L. (2020) Oligosaccharide binding and thermostability of two related aa9 lytic polysaccharide monoxygenases. *Biochemistry* **59**, 3347–3358
34. Laurent, C., Sun, P., Scheiblbrandner, S., Csarman, F., Cannazza, P., Frommhagen, M., van Berkel, W. J. H., Oostenbrink, C., Kabel, M. A., and Ludwig, R. (2019) Influence of lytic polysaccharide monoxygenase active site segments on activity and affinity. *Int. J. Mol. Sci.* **20**, 6219
35. Chen, C., Chen, J., Geng, Z., Wang, M., Liu, N., and Li, D. (2018) Regioselectivity of oxidation by a polysaccharide monoxygenase from *Chaetomium thermophilum*. *Biotechnol. Biofuels* **11**, 155
36. Frandsen, K. E. H., Poulsen, J.-C. N., Tandrup, T., and Lo Leggio, L. (2017) Unliganded and substrate bound structures of the cellooligosaccharide active lytic polysaccharide monoxygenase LsAA9A at low pH. *Carbohydr. Res.* **448**, 187–190
37. Span, E. A., Suess, D. L. M., Deller, M. C., Britt, R. D., and Marletta, M. A. (2017) The role of the secondary coordination sphere in a fungal polysaccharide monoxygenase. *ACS Chem. Biol.* **12**, 1095–1103
38. Wu, M., Beckham, G. T., Larsson, A. M., Ishida, T., Kim, S., Payne, C. M., Himmel, M. E., Crowley, M. F., Horn, S. J., Westereng, B., Igarashi, K., Samejima, M., Stahlberg, J., Eijsink, V. G., and Sandgren, M. (2013) Crystal structure and computational characterization of the lytic polysaccharide monoxygenase GH61D from the Basidiomycota fungus *Phanerochaete chrysosporium*. *J. Biol. Chem.* **288**, 12828–12839
39. Westereng, B., Ishida, T., Vaaje-Kolstad, G., Wu, M., Eijsink, V. G., Igarashi, K., Samejima, M., Stahlberg, J., Horn, S. J., and Sandgren, M. (2011) The putative endoglucanase PcGH61D from *Phanerochaete chrysosporium* is a metal-dependent oxidative enzyme that cleaves cellulose. *PLoS One* **6**, e27807
40. Lo Leggio, L., Weihe, C. D., Poulsen, J.-C. N., Sweeney, M., Rasmussen, F., Lin, J., De Maria, L., and Wogulis, M. (2018) Structure of a lytic polysaccharide monoxygenase from *Aspergillus fumigatus* and an engineered thermostable variant. *Carbohydr. Res.* **469**, 55–59
41. Fry, S. C., York, W. S., Albersheim, P., Darvill, A., Hayashi, T., Joseleau, J.-P., Kato, Y., Lorences, E. P., MacLachlan, G. A., McNeil, M., Mort, A. J., Grant Reid, J. S., Seitz, H. U., Selvendran, R. R., Voragen, A. G. J., et al. (1993) An unambiguous nomenclature for xyloglucan-derived oligosaccharides. *Physiol. Plant.* **89**, 1–3
42. Waterhouse, A., Bertoni, M., Bienert, S., Tauriello, G., Gumienny, R., Heer, F. T., de Beer, T. A. P., Rempfer, C., Bordoli, L., Lepore, R., and Schwede, T. (2018) SWISS-MODEL: homology modelling of protein structures and complexes. *Nucleic Acids Res.* **46**, W296–W303
43. Jurak, E., Suzuki, H., van Erven, G., Gandier, J. A., Wong, P., Chan, K., Ho, C. Y., Gong, Y., Tillier, E., Rosso, M. N., Kabel, M. A., Miyauchi, S., and Master, E. R. (2018) Dynamics of the *Phanerochaete carnosus* transcriptome during growth on aspen and spruce. *BMC Genomics* **19**, 815
44. Jagadeeswaran, G., Gainey, L., Prade, R., and Mort, A. J. (2016) A family of AA9 lytic polysaccharide monoxygenases in *Aspergillus nidulans* is differentially regulated by multiple substrates and at least one is active on cellulose and xyloglucan. *Appl. Microbiol. Biotechnol.* **100**, 4535–4547
45. Jagadeeswaran, G., Gainey, L., and Mort, A. J. (2018) An AA9-LPMO containing a CBM1 domain in *Aspergillus nidulans* is active on cellulose and cleaves cello-oligosaccharides. *AMB Express* **8**, 171
46. Monclaro, A. V., Petrovic, D. M., Alves, G. S. C., Costa, M. M. C., Midorikawa, G. E. O., Miller, R. N. G., Filho, E. X. F., Eijsink, V. G. H., and Varnai, A. (2020) Characterization of two family AA9 LPMOs from *Aspergillus tamarii* with distinct activities on xyloglucan reveals structural differences linked to cleavage specificity. *PLoS One* **15**, e0235642
47. Haon, M., Grisel, S., Navarro, D., Gruet, A., Berrin, J. G., and Bignon, C. (2015) Recombinant protein production facility for fungal biomass-degrading enzymes using the yeast *Pichia pastoris*. *Front. Microbiol.* **6**, 1002
48. Kittl, R., Kracher, D., Burgstaller, D., Haltrich, D., and Ludwig, R. (2012) Production of four *Neurospora crassa* lytic polysaccharide monoxygenases in *Pichia pastoris* monitored by a fluorimetric assay. *Biotechnol. Biofuels* **5**, 79
49. Breslmayr, E., Hanžek, M., Hanrahan, A., Leitner, C., Kittl, R., Šantek, B., Oostenbrink, C., and Ludwig, R. (2018) A fast and sensitive activity assay for lytic polysaccharide monoxygenase. *Biotechnol. Biofuels* **11**, 79
50. McGregor, N., Morar, M., Fenger, T. H., Stogios, P., Lenfant, N., Yin, V., Xu, X., Evdokimova, E., Cui, H., Henrissat, B., Savchenko, A., and Brumer, H. (2016) Structure-function analysis of a mixed-linkage beta-glucanase/xyloglucanase from the key ruminal bacteroidetes *Prevotella bryantii* B(1)4. *J. Biol. Chem.* **291**, 1175–1197
51. Westereng, B., Agger, J. W., Horn, S. J., Vaaje-Kolstad, G., Aachmann, F. L., Stenstrom, Y. H., and Eijsink, V. G. (2013) Efficient separation of oxidized cello-oligosaccharides generated by cellulose degrading lytic polysaccharide monoxygenases. *J. Chromatogr. A* **1271**, 144–152
52. Kelley, L. A., Mezulis, S., Yates, C. M., Wass, M. N., and Sternberg, M. J. (2015) The Phyre2 web portal for protein modeling, prediction and analysis. *Nat. Protoc.* **10**, 845–858