Featuring work from the research group of Professor Bert F. Sels and collaborators, Center for Surface Chemistry and Catalysis, KU Leuven, Belgium.

Chemicals from lignin: an interplay of lignocellulose fractionation, depolymerisation, and upgrading

A critical review of past and recent developments in the production of chemicals from lignin, focusing on lignocellulose fractionation, lignin depolymerisation, and upgrading.

As featured in:

See B. F. Sels *et al.*, *Chem. Soc. Rev.*, 2018, **47**, 852.

ROYAL SOCIETY OF CHEMISTRY

rsc.li/chem-soc-rev

Registered charity number: 207890

Chem Soc Rev

REVIEW ARTICLE

View Article Online
View Journal | View Issue

Cite this: *Chem. Soc. Rev.*, 2018, **47**, 852

Chemicals from lignin: an interplay of lignocellulose fractionation, depolymerisation, and upgrading†

W. Schutyser, ‡[ab] T. Renders, ‡[a] S. Van den Bosch, [a] S.-F. Koelewijn, [a] G. T. Beckham [b] and B. F. Sels *[a]

In pursuit of more sustainable and competitive biorefineries, the effective valorisation of lignin is key. An alluring opportunity is the exploitation of lignin as a resource for chemicals. Three technological biorefinery aspects will determine the realisation of a successful lignin-to-chemicals valorisation chain, namely (i) lignocellulose fractionation, (ii) lignin depolymerisation, and (iii) upgrading towards targeted chemicals. This review provides a summary and perspective of the extensive research that has been devoted to each of these three interconnected biorefinery aspects, ranging from industrially well-established techniques to the latest cutting edge innovations. To navigate the reader through the overwhelming collection of literature on each topic, distinct strategies/topics were delineated and summarised in comprehensive overview figures. Upon closer inspection, conceptual principles arise that rationalise the success of certain methodologies, and more importantly, can guide future research to further expand the portfolio of promising technologies. When targeting chemicals, a key objective during the fractionation and depolymerisation stage is to minimise lignin condensation (*i.e.* formation of resistive carbon–carbon linkages). During fractionation, this can be achieved by either (i) preserving the (native) lignin structure or (ii) by tolerating depolymerisation of the lignin polymer but preventing condensation through chemical quenching or physical removal of reactive intermediates. The latter strategy is also commonly applied in the lignin depolymerisation stage, while an alternative approach is to augment the relative rate of depolymerisation *vs.* condensation by enhancing the reactivity of the lignin structure towards depolymerisation. Finally, because depolymerised lignins often consist of a complex mixture of various compounds, upgrading of the raw product mixture through convergent transformations embodies a promising approach to decrease the complexity. This particular upgrading approach is termed funneling, and includes both chemocatalytic and biological strategies.

Received 1st August 2017

DOI: 10.1039/c7cs00566k

rsc.li/chem-soc-rev

[a] *Center for Surface Chemistry and Catalysis, KU Leuven, Celestijnenlaan 200F, 3001 Heverlee, Belgium. E-mail: bert.sels@biw.kuleuven.be*
[b] *National Bioenergy Center, National Renewable Energy Laboratory, 15013 Denver West Parkway, Golden, Colorado 80401, USA*

† Electronic supplementary information (ESI) available. See DOI: 10.1039/c7cs00566k

‡ Authors contributed equally to this work.

W. Schutyser

Wouter Schutyser obtained his PhD degree in Bioscience Engineering (Catalytic Technology) in 2015 at KU Leuven under the guidance of Prof. Bert Sels, in which he studied the reductive upgrading of lignin. In 2016–2017, he did postdoctoral work in the group of Dr Gregg Beckham at NREL, examining lignin depolymerisation through oxidative pathways. He is currently an FWO postdoctoral researcher in the group of Prof. Bert Sels at KU Leuven, where he continues to explore the catalytic valorization of lignin.

T. Renders

Tom Renders obtained his MSc in Bioscience Engineering (Catalytic Technology) at KU Leuven in 2014. He did his master thesis at the Centre for Surface Chemistry and Catalysis under the guidance of Prof. Bert F. Sels, where he investigated the conversion of polybutadiene by olefin metathesis. As an FWO-funded fellow, he is currently doing a PhD in the same research group. His research focuses on lignocellulose biorefining, with emphasis on lignin valorisation towards chemicals.

1. Introduction

One of the foremost challenges in the 21st century is the replacement of fossil resources by more sustainable alternatives.[1,2] A viable substitute should at least be renewable, CO_2-neutral, widely available, and should not compete with food production. Inedible plant biomass in the form of lignocellulose is one of the few resources that meets these essential criteria. However, in contrast to petroleum, lignocellulose is a highly oxygenated, solid, and heterogeneous substance.[3–5] It is mainly composed of three highly functional biopolymers, *viz.* cellulose, hemicellulose, and lignin, forming a composite material which is resistant to (bio)chemical conversion, a phenomenon commonly known as biomass recalcitrance.[6,7]

Unfortunately, there is no single or universal strategy to cope with these inherent obstacles of plant biomass. Over the years, different biorefinery strategies have been proposed, each with their own advantages and limitations. A classic example of a lignocellulosic biorefinery is a pulp or paper mill, which targets the manufacture of high quality pulps for the paper and cardboard industry.[8] Another well-known, emerging illustration is the fermentative production of bio-ethanol from lignocellulosic carbohydrates.[9,10] It is clear that for these examples, the primary products are derived from the carbohydrate fraction, whereas the lignin portion is mostly regarded as a low-value by-product or a cheap energy source. However, the economic viability and environmental sustainability of a biorefinery can be significantly increased if the valorisation of lignin, the largest renewable source of aromatics, becomes fully integrated as well.[11–14]

S. Van den Bosch

Sander Van den Bosch obtained his MSc in Bioscience Engineering at KU Leuven in 2013. He did his master thesis at the Centre for Surface Chemistry and Catalysis under the guidance of Prof. Bert F. Sels, where he investigated the catalytic valorisation of lignin to fine chemicals. As an IWT-funded fellow, he is currently finishing a PhD in the same research group. His research focuses on the valorisation of lignin in the biorefinery.

S.-F. Koelewijn

Steven-Friso Koelewijn obtained his MSc degree in Bioscience Engineering (Catalytic Technology) at KU Leuven in 2013. He did his master's thesis at the Centre for Surface Chemistry and Catalysis under the guidance of Prof. Bert F. Sels, where he explored the catalytic synthesis of renewable bisphenols by hyperbranched polymers. He is currently in the progress of finishing his PhD degree in the same group. In general his research focuses on the valorization of lignin-derived aromatic monomers. More specifically, his research interests include green chemistry, toxicological chemistry, polymer chemistry, which converge in the concept of 'safety-by-design'.

G. T. Beckham

Gregg T. Beckham is a Group Leader and Senior Engineer at NREL. He received his PhD in Chemical Engineering at MIT in 2007. He currently leads and works with an interdisciplinary team of biologists, chemists, and engineers at the National Renewable Energy Laboratory on conversion of biomass to fuels, chemicals, and materials including in metabolic engineering, fermentation, separations, catalysis, biopolymer and carbon fiber production, and lignin valorisation.

B. F. Sels

Bert F. Sels, currently full professor at KU Leuven, obtained his PhD degree in 2000 in the field of heterogeneous oxidation catalysis. He was awarded the DSM Chemistry Award in 2000, the Incentive Award by the Belgian Chemical Society in 2005 and the Green Chemistry Award in 2015. He is currently director of the Centre for Surface Chemistry and Catalysis, designing heterogeneous catalysts for future challenges in industrial organic and environmental catalysis. His expertise includes heterogeneous catalysis in bio-refineries, design of hierarchical zeolites and carbons and the spectroscopic and kinetic study of active sites for small-molecule activation. He authored about 280 peer reviewed papers and filed 25 patents.

Historically, a major thrust for lignin valorisation has been generated by research focusing on macromolecular applications. This research domain (*viz.* lignin-to-materials) remains still prominent today, and has brought forth a wide array of promising applications, ranging from glues, adsorbents, and polymer composites to raw material for carbon nanofibers.[11,15–19] More recently, the utilisation of lignin as a feedstock for the production of bio-based chemicals is receiving increasing attention. Efficient lignin disassembly, followed by targeted upgrading, could provide access to a wide variety of chemicals and fuel components,[11–13,18,20–24] complementary to a well-developed carbohydrate platform.[25–28] This biorefinery target, *viz.* lignin-to-chemicals, forms the subject of the following review.

The successful conversion of lignin to value-added chemicals – from a technology and chemistry point of view – is mainly determined by an interplay of three important biorefinery aspects: (i) lignocellulose fractionation, (ii) lignin depolymerisation, and finally (iii) upgrading towards desired chemicals. Over the past few decades, these three aspects have been the subject of extensive research, both in academia and industry. The result is a complex and overwhelming assortment of available technologies.[23,24]

This review attempts to provide a comprehensive overview of the research that has been devoted to the exploitation of lignin as a feedstock for chemicals, and is structured into three main (interconnected) sections, *viz.* biomass fractionation (Section 4), lignin depolymerisation (Section 5), and upgrading (Section 6). Each section is accompanied by intelligible graphical overviews of the current state-of-the-art, which we hope will make the review easily accessible to a wide audience. Furthermore, for each biorefinery aspect, a perspective is presented of the underlying fundamental principles that have enabled the success of a given unit operation *en route* to lignin-derived chemicals. Preceding the three main sections (Sections 4–6), a brief introduction on lignocellulosic biomass is included (Section 2), followed by an overview of the different types of lignin chemistry (Section 3), which form the foundation of many fractionation and depolymerisation technologies.

2. Lignocellulosic biomass: a brief introduction

2.1 Lignocellulose composition

Lignocellulose is the major structural component of plants,[3,4] and is by far the most abundant type of terrestrial biomass.[29] It is a composite material mainly consisting of cellulose (40–60%), hemicellulose (10–40%), and lignin (15–30%);[3,4] and can be found in both woody (*e.g.* pine, poplar, birch) and herbaceous biomass (*e.g.* switchgrass, miscanthus, corn stover). The cellulose portion is exclusively composed of glucose units, which are linked in a linear fashion *via* β-1,4-glycosidic bonds.[30–33] The resulting polymer chains can have a polymerisation degree of up to 10 000 units.[30] These chains interact with each other *via* hydrogen bonds and van der Waals forces, eventually giving rise to rigid, semi-crystalline cellulose fibres.[30–35] Due to these strong interaction forces, cellulose fibres are insoluble in most conventional solvents, including water.

The second carbohydrate polymer in lignocellulosic biomass is hemicellulose, which represents a family of branched carbohydrate polymers containing both pentoses (*e.g.* xylose, arabinose) and hexoses (*e.g.* galactose, glucose, mannose).[36–38] Uronic acids (*e.g.* glucuronic acid) and acetyl moieties are often present as side-chain groups.[36–40] The chemical composition of hemicellulose can strongly vary, depending on the botanical origin of the biomass. The degree of polymerisation is generally lower, in the range of 50–300 units.[40,41] Unlike cellulose, hemicellulose is an amorphous biopolymer which is therefore more easily solubilised, and is more prone to chemical attack. The total carbohydrate fraction is often referred to as holocellulose, and includes cellulose, hemicellulose, as well as other (minor) carbohydrate bio-polymers such as pectins. Pectins account for only a small fraction of the carbohydrates in grasses, but contribute significantly to the biomass recalcitrance.[42,43]

The focal point of this review is the valorisation of lignin, the third and most complex constituent of lignocellulosic biomass. Lignin is defined as an irregular, oxygenated *p*-propylphenol polymer, formed by free radical polymerisation of monolignols (Fig. 1) in the plant cell wall,[44–46] and therefore has been referred to as supramolecular self-assembled chaos.[47] It provides rigidity to the plant cell wall as well as resistance to microbial attack,[3,48] and thus contributes to biomass recalcitrance.[3,7] Lignin biosynthesis is initiated by the phenylpropanoid pathway, a multi-enzyme biochemical network wherein phenylalanine (and tyrosine, in grasses)[49,50] is converted into the main lignin building blocks: *p*-coumaryl alcohol, coniferyl alcohol, and sinapyl alcohol (Fig. 1).[46,51] These building blocks, or monolignols, differ in the amount of methoxy groups on the phenolic nucleus, which are commonly abbreviated as H (*p*-hydroxyphenyl), G (guaiacyl), and S (syringyl). The relative distribution of phenolic nuclei in lignin strongly differs between plant species. In general, softwood lignin (*e.g.* pine, spruce) exclusively contains G units, whereas hardwood lignin (*e.g.* birch, poplar, eucalyptus) is composed of both G and S units. Lignin from herbaceous biomass contains all three units, although the H-content is generally low (<5%).[51] Next to the main building blocks, lignin can also incorporate substantial amounts of other (phenolic) compounds such as hydroxycinnamates (*p*-coumarate (**1**), ferulate (**2**), *etc.*),[51,52] *p*-hydroxybenzoate (**3**),[53–55] tricin (**4**),[56–59] acetate,[60,61] and other products from incomplete monolignol biosynthesis (Fig. 2).[51]

Following their biosynthesis in the cytosol,[62] the monolignols are translocated to the cell wall where lignification (radical polymerisation) takes place.[44,63] Lignification is initiated by laccase and peroxidase enzymes that oxidise the phenolic OH-group, resulting in phenoxy radicals.[44,45,51,63] The actual polymerisation reactions occur through the combination (termination) of two radicals. Because of the monolignol's conjugated π-system, various inter-unit linkages can be formed (Fig. 1), including both ether and carbon–carbon bonds.[44–46] The most abundant and well-known linkage type is the β-O-4 ether motif. Together with the α-O-4 linkage (in phenylcoumaran and dibenzodioxocin structures, Fig. 1), these ether bonds are most easily cleaved and they are therefore often the primary

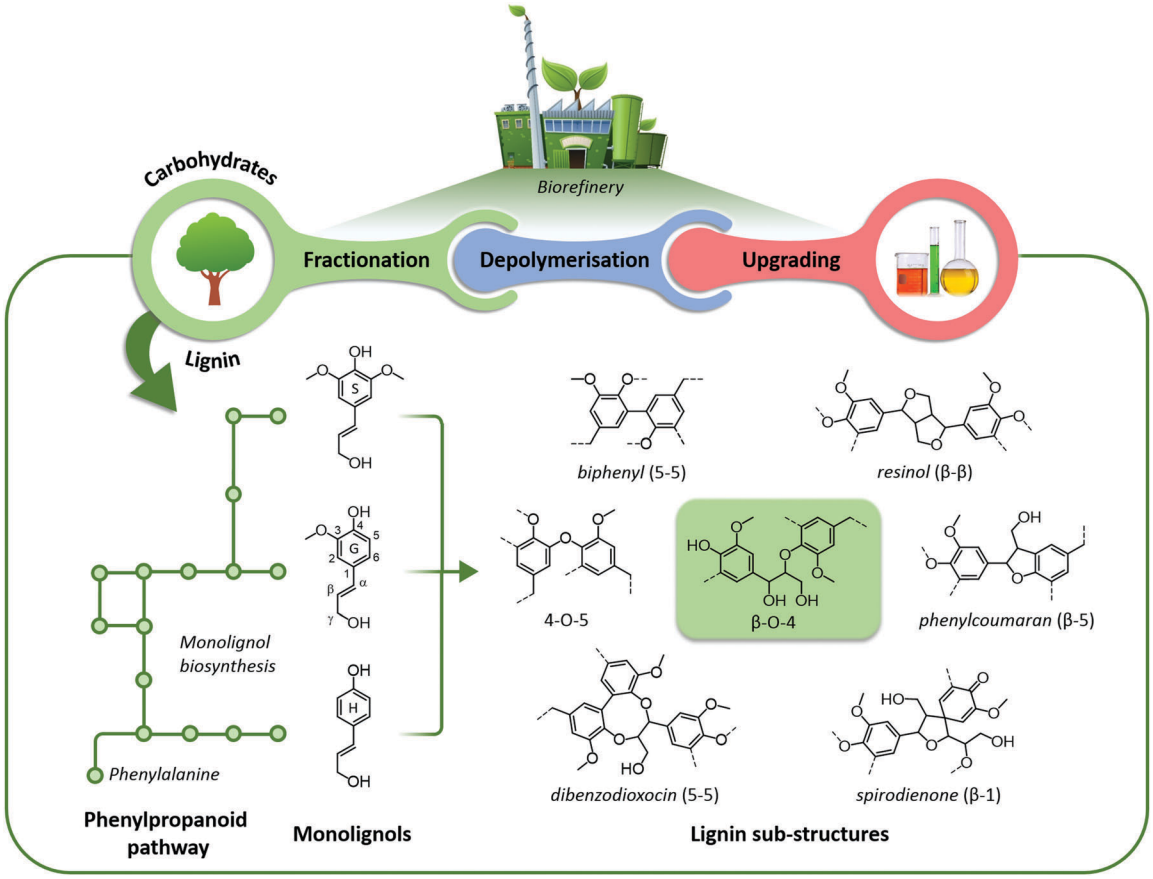

Fig. 1 The biosynthesis of lignin starts with the synthesis of monolignols through the phenylpropanoid pathway, followed by translocation to the cell wall where radical polymerisation takes place, leading to various ether and carbon–carbon inter-unit bonds. The successful utilisation of this complex bio-polymer as a feedstock for chemicals is governed by an interplay of (i) the biomass fractionation method, (ii) the lignin depolymerisation technology, and (iii) subsequent upgrading towards targeted chemicals. These three interconnected stages form the backbone of the following review.

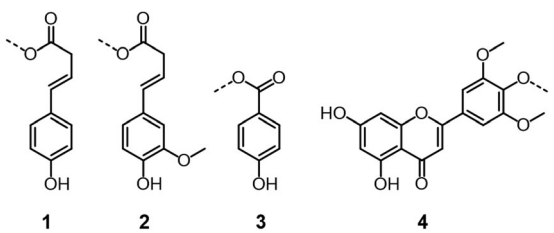

Fig. 2 Examples of other lignin building blocks: *p*-coumarate (**1**, in herbaceous crops), ferulate (**2**, in herbaceous crops), *p*-hydroxybenzoate (**3**, *e.g.* in poplar and palm), and tricin (**4**, in herbaceous crops).

target of lignin depolymerisation methods.[64,65] In contrast to carbohydrate polymers, lignin also contains a substantial fraction of carbon–carbon inter-unit bonds, such as 5-5 (in dibenzodioxocin and biphenyl), β-5 (in phenylcoumaran), β-1 (in spirodienone) and β-β (in resinol) connections (Fig. 1). The relative amount of carbon–carbon inter-unit bonds in native lignin is strongly governed by the monolignol distribution (*i.e.* S/G/H ratio). This relationship can be elucidated by the fact that methoxy-substituted *ortho*-positions cannot participate in the formation of 5-5 and β-5 carbon–carbon bonds. Therefore, lignin that is primarily composed of S-units (hardwood) contains a lower fraction of carbon–carbon bonds than lignin composed of G-units (softwood).[62,66] These differences can have significant consequences for biomass delignification and lignin depolymerisation (*vide infra*).

The molecular weight of native lignin is difficult to appoint, due to structural changes occurring during its isolation. However, close-to-native lignins have been reported to exhibit number average molecular weights in the range of 2500–10 000 g mol^{-1}.[67] Native lignin is strongly associated with the hemicellulose fraction, forming an amorphous matrix which encapsulates and supports the rigid cellulose fibres. In this matrix, lignin is covalently cross-linked with hemicellulose, giving rise to lignin–carbohydrate complexes (LCCs).[68–70] The extent of lignification (*viz.* portion of lignin in the biomass) depends on the botanical origin of a plant. As a rule of thumb, softwoods contain the highest amount of lignin (21–29 wt%), followed by hardwoods (18–25 wt%) and herbaceous crops (15–24 wt%).[71–73] Variation furthermore occurs between different tissues, cell types, regions in the cell wall, and between different life stages of a cell.[5,31] Finally, lignification is influenced by physical stress.[5,74] These factors can alter the monolignol distribution as well, thereby affecting the lignin chemistry and structure.[74]

2.2 The biorefinery: from recalcitrant resource to valuable products

To reduce the complexity, heterogeneity, and recalcitrance of lignocellulosic biomass, fractionation is often performed as a primary step in the biorefinery. With respect to lignin, most conventional fractionation techniques aim for its recovery as a solid product. As will be discussed in Section 4, a multitude of isolated lignins is nowadays available, each with their own structural and chemical characteristics. Commercial or industrial lignins are frequently denoted as technical lignins (*e.g.* kraft lignin, lignosulfonates, soda lignin, organosolv lignin).[75] Some of these (technical) lignins, in particular lignosulfonates,[76] can be utilised in macromolecular applications, for example as emulsifier, binding agent, rheology modifier, battery electrode, carbon fibre precursor, in polymer formulations, *etc.* The interested reader is referred to more dedicated literature on this topic (*viz.* lignin-to-materials).[15–17,19,76–80]

An auspicious and complementary valorisation opportunity is to apply lignin as a renewable resource for low M_w chemicals (lignin-to-chemicals, the focal point of this review). Thereto, adequate depolymerisation (Section 5) followed by chemo- or biocatalytic upgrading (Section 6) is needed to construct a successful lignin valorisation chain. Fractionation, depolymerisation, and upgrading are most often thought of as physically separated, consecutive processes (Fig. 3). As an (hypothetical) example of this traditional model: one way to generate a fuel-compatible liquid encompasses Kraft pulping (fractionation); followed by lignin pyrolysis (depolymerisation), and subsequent hydrodeoxygenation (upgrading), eventually yielding cycloalkanes. Alternatively, two processing steps can also be performed simultaneously, in a single operation. Within this context, reductive catalytic fractionation (RCF) is an emerging technology that combines fractionation (similar to organosolv pulping) and reductive depolymerisation–stabilisation, leading directly to a depolymerised lignin oil (Section 5.1.1).[64] This approach mitigates the issue of lignin degradation, a problem frequently encountered during traditional lignin isolation techniques. In analogy, processes exist wherein depolymerisation is combined with upgrading, for instance during bifunctional hydroprocessing of lignin towards cycloalkanes (Section 5.2.1).

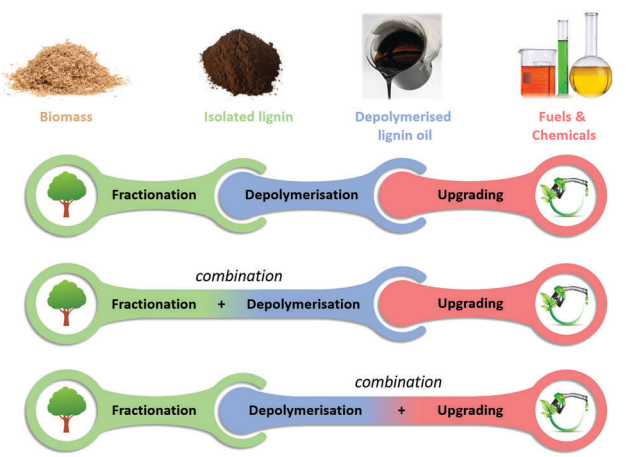

Fig. 3 A lignin-to-chemicals valorisation chain comprises (i) biomass fractionation, (ii) lignin depolymerisation, and (iii) upgrading. These operations are mostly executed as physically separated, consecutive processes. However, combinatorial approaches exist wherein two steps occur simultaneously in a single process.

3. Lignin chemistry

In any biomass or lignin processing method, the lignin structure is altered through a combination of de- and repolymerisation reactions. The outcome strongly depends on the underlying mechanism of the processing method, *viz.* (i) base-catalysed, (ii) acid-catalysed, (iii) reductive, (iv) oxidative, or (v) thermal. This section provides an overview of the possible chemical reactions during lignin or lignocellulose processing, with focus on the fate of β-O-4 motifs. These reactive linkages largely dictate the lignin reactivity as they are the most abundant and overall the most reactive inter-unit linkages, and often form the primary target in biomass fractionation and depolymerisation.

3.1 Base-catalysed lignin chemistry

Lignin is rather insoluble in water owing to its medium-polarity, though, its water solubility is significantly higher in alkaline media due to deprotonation of phenolic OH-groups. Alkaline conditions are therefore frequently applied in processes which aim to extract lignin from the biomass (*i.e.* delignification), for instance in traditional pulping processes (Section 4.1).[8,81–83] Alkaline media enable the cleavage of lignin–carbohydrate bonds, the fragmentation of lignin *via* cleavage of the β-O-4 motifs, the solubilisation of the resulting fragments, and eventually lignin degradation/repolymerisation.[83–86]

Typical base-catalysed reactions on the most prevalent lignin sub-structure, the β-O-4 motif, are illustrated in Fig. 4. The reactivity of the β-O-4 linkages strongly depends on the type of units: a distinction is made between phenolic units (free phenolic OH-groups) and non-phenolic units (etherified phenolic OH-groups).[85] Cleavage of the β-O-4 bonds in non-phenolic units (**5**) has been postulated to proceed relatively slowly through the base-catalysed formation of an epoxide intermediate (**7**).[82–84,87,88] This reaction generates phenolic units (**6**), which are subsequently depolymerised more easily. First, the phenolic unit is transformed into a quinone methide (**8**), provided that a suitable leaving group (*e.g.* –OH, –OR) is present at the α-position (Fig. 4).[85,86,88,89] The quinone methide – a pivotal point in alkaline lignin chemistry – is prone to undergo nucleophilic attack, owing to its tendency to restore the aromaticity. In presence of a strong nucleophilic anion such as HS^- (Kraft pulping), the prevailing pathway is the cleavage of the β-O-4 motif *via* the formation of episulfide intermediates (**11**).[82,85,86,89] These sulfur-containing intermediates may undergo a variety of subsequent reactions, yielding compounds such as coniferyl alcohol which are prone to degradation and repolymerisation.[21,88] In a competing reaction, the quinone methide can react with an *in situ* formed lignin nucleophile, leading to repolymerisation through formation of a carbon–carbon bond. An alternative (third) pathway to restore the

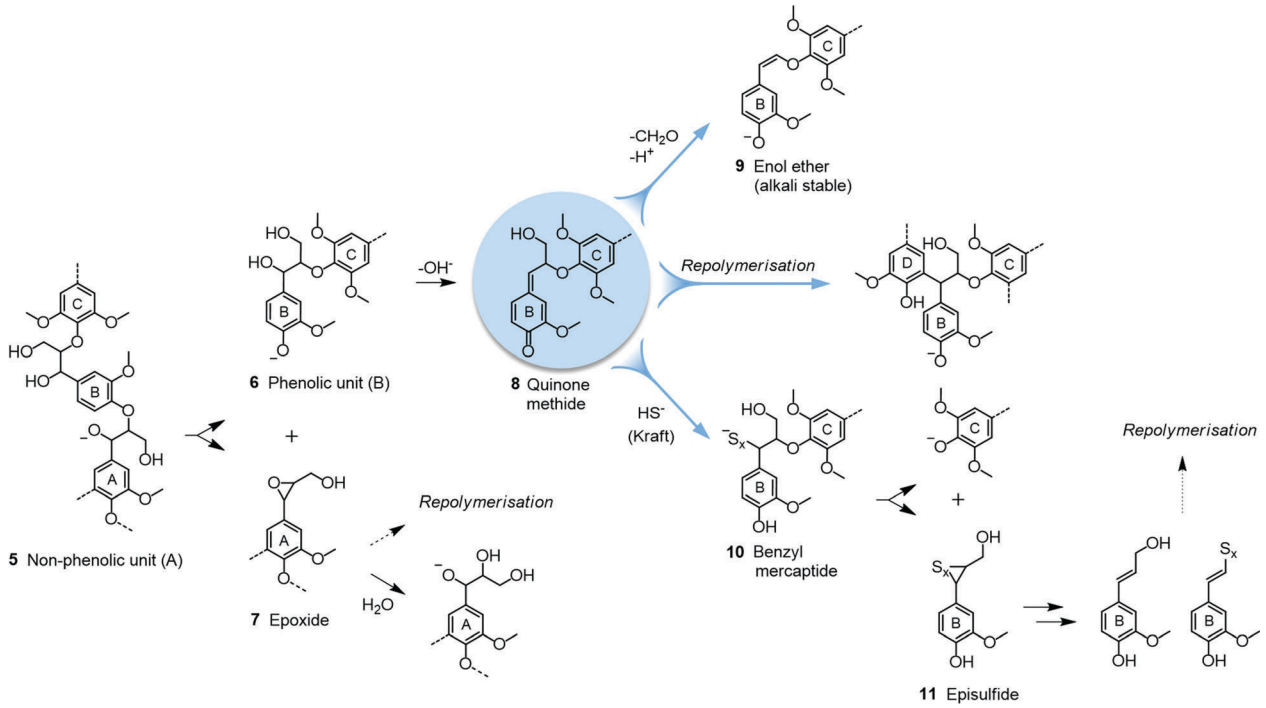

Fig. 4 Alkaline lignin chemistry. For clarity, phenolic rings are labelled with A–D.

aromaticity proceeds through the removal of the terminal γ-CH_2OH group *via* a retro-aldol type reaction.[85,86,89] The outcome of this route is the formation of an alkali-stable enol ether motif (**9**) and formaldehyde. This pathway occurs more frequently in absence of a strong nucleophile, as is the case in soda pulping.[21] Consequently, ether bonds are cleaved less efficiently. The as-formed formaldehyde can induce repolymerisation *via* formaldehyde-phenol type condensation.[21,85,89]

3.2 Acid-catalysed lignin chemistry

Acidic conditions are known to promote hydrolysis of ether bonds in the carbohydrate polymers, and are therefore often applied to depolymerise and solubilise the hemicellulose and/or cellulose fraction (Section 4.2). Unlike alkaline conditions, acidic environments do not necessarily promote the solubilisation and extraction of lignin.[90,91] Nevertheless, acidic media affect the lignin structure by facilitating both depolymerisation (*i.e.* acidolysis) and repolymerisation.[91,92]

The most prominent event in acid-catalysed lignin chemistry is the cleavage of β-O-4 ether bonds,[92–96] depicted in Fig. 5. The first step of β-O-4 acidolysis is the formation of a benzylic carbenium ion (**12**) by removal of the OH-group on the α-position. The intermediate carbenium ion can transform into two enol-ether structures (**13a** and **13b**), *viz.* with or without cleavage of the C_β–C_γ bond and concurrent formation of formaldehyde (Fig. 5).[92–96] The prevailing pathway depends on the applied acid. For instance, the formation of intermediate **13b** predominates when utilising H_2SO_4, while **13a** is the main enol ether in case of HCl or HBr.[93–95] Subsequent hydrolysis of the acid-labile enol ethers yields (i) C_2-aldehyde-substituted phenolics (**15**) and (ii) C_3-ketone-substituted phenolics (**14**).[92–98] The latter are referred to as Hibbert's ketones.[99–101] Hibbert's ketones, together with carbenium ions and C_2-aldehyde-substituted phenolics participate in a complex network of repolymerisation reactions, eventually leading to a condensed lignin polymer.[93,97,102]

3.3 Reductive lignin chemistry

Reductive conditions have been frequently applied in depolymerisation processes of (isolated) lignins. To this end, a redox catalyst in combination with H_2 or a H-donor is essential. Reductive processing primarily targets the inter-unit ether bonds (β-O-4, α-O-4) and side-chain hydroxyl groups. Different pathways and mechanisms have been proposed,[103–107] and the net result in all cases is: (i) hydrogenolysis of ether bonds, (ii) removal of benzylic OH-groups (OH_α), and (iii) possible removal of OH_γ-groups (Fig. 6). These primary reactions lead to the formation of substituted methoxyphenols (**16**) and small oligomeric fragments. Depending on the processing conditions and catalyst used, additional hydrogenation/hydrogenolysis can take place (secondary reactions), for instance leading to cyclohexanols or cycloalkanes. Noteworthy is the ability of these reductive catalytic systems to quench reactive functional groups that are prone to condensation, such as alkenyl and carbonyl groups. Hence, reductive conditions can avoid repolymerisation (at least to a certain extent), in contrast to alkaline or acidic media. However, on the downside is the inability of most reductive processes to cleave carbon–carbon linkages. Consequently, the degree of depolymerisation is typically linked to the relative amount of cleavable inter-unit ether bonds present in the lignin polymer before processing.

3.4 Oxidative lignin chemistry

Oxidation of lignin has traditionally been performed in pulp bleaching, with the aim of producing high-quality paper products.

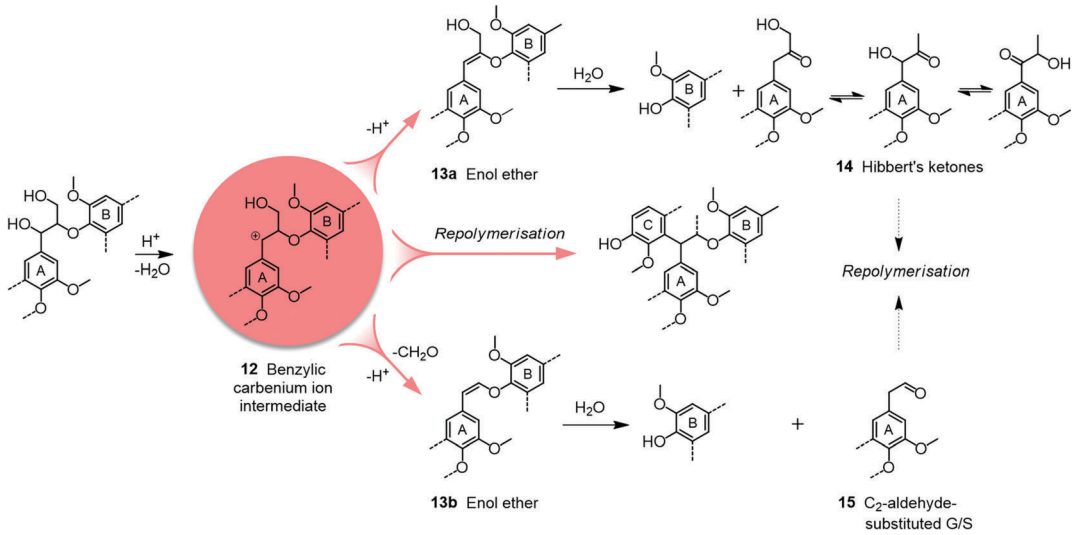

Fig. 5 Acid-catalysed lignin chemistry.

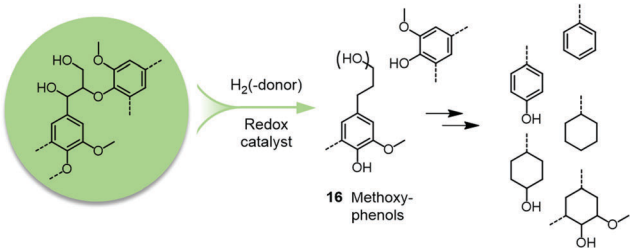

Fig. 6 Reductive lignin chemistry.

During bleaching, the intensely coloured residual lignin (2–6 wt%) is degraded and/or removed, which increases the brightness of the pulp.[48] Various oxidants are available, such as chlorine, chlorine dioxide, oxygen, hydrogen peroxide, ozone, and peroxyacids.[48,108,109] The mechanism of lignin oxidation by these oxidants was extensively discussed by Gierer *et al.*[109] and more recently reviewed by Ma *et al.*[108] Most oxidation mechanisms are initiated by electrophilic reactions, *i.e.* attack of electrophilic species, such as Cl^+ (from chlorine), OH^+ (from peroxyacids), or oxygen, on sites of high electron density, such as *ortho*, *para*, or C_β positions in lignin.[109] Oxygen is a particularly interesting oxidant, as it is inexpensive, readily available, and produces water as main side-product. However, since oxygen is a weak oxidising agent in its normal state, oxidation under oxygen atmosphere (aerobic oxidation) often requires alkaline conditions to ionise the free phenolic hydroxyl groups in lignin.[108]

Alkaline aerobic lignin oxidation proceeds through a radical reaction mechanism.[108,109] It is initiated by oxidation of the phenolate ions into phenoxy radicals. The exact mechanism of lignin depolymerisation under alkaline aerobic oxidation conditions is not yet fully understood, but the most commonly accepted pathways are depicted in Fig. S1 in the ESI†. According to Gierer *et al.*, oxygen adds to the phenoxy radicals in *ortho*, *para*, or C_β position, leading to the formation of peroxy anions.[109] The peroxy anions subsequently transform through several routes, resulting in either: (i) cleavage of the C_α–C_β bond, generating phenolic aldehydes (**17**), (ii) C_4–C_α cleavage, leading to the formation of *p*-quinones (**19**), (iii) formation of oxirane structures, and (iv) cleavage of the aromatic ring, yielding muconic acid derivatives (**20**).[108–110] In contrast to Gierer *et al.*, Tarabanko *et al.* have suggested that alkaline aerobic lignin oxidation does not involve oxygen addition, but proceeds through a second oxidation (electron abstraction) of the phenoxy radicals, yielding a cinnamaldehyde-like intermediate, followed by disruption of the C_α–C_β bond through retro-aldol cleavage.[23,111–113]

In summary, depending on the pathway that is followed, either the side-chain is fragmented (C_α–C_β or C_4–C_α bond cleavage) and the aromaticity is retained, or the aromatic ring is disrupted, yielding aliphatic carboxylic acids (Fig. 7). The aromatic structures obtained through C_α–C_β or C_4–C_α bond cleavage are however not stable under alkaline oxidation conditions, and they can be further converted to aliphatic carboxylic acids (**20**) through aromatic ring cleavage.[108,109,114] So in contrast to reductive processing, the generated phenolic products are not generally as stable under oxidative conditions. According to the proposed mechanisms, oxidative lignin depolymerisation mostly proceeds

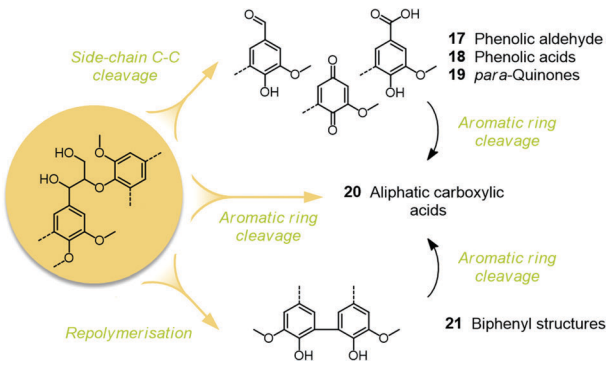

Fig. 7 Oxidative lignin chemistry. Proposed oxidation mechanisms are depicted in Fig. S1 (ESI†).

through carbon–carbon bond cleavage instead of ether-bond cleavage, as in acid-, base- or reductive lignin depolymerisation. Because a radical reaction mechanism is followed, condensation of lignin fragments through radical coupling also takes place, yielding biphenyl structures (**21**).[108]

3.5 Thermal lignin chemistry

Given the significant research and development on pyrolysis in the biomass conversion community, the thermal degradation mechanisms of lignin and lignin model compounds have been studied quite extensively to date, but mechanistic consensus has not yet been fully achieved. We note that the studies in this area are far too numerous to comprehensively review, and readers are referred to more detailed treatments of this topic.[23,115–118] The primary mechanistic question in thermal degradation of lignin at pyrolysis conditions, which remains unanswered in a definitive manner, is whether C–O and C–C bond cleavage reactions proceed *via* homolytic bond scission reactions to generate radicals that further recombine, or if the bond cleavage reactions are concerted in nature. As an illustrative example, thermal cleavage of the β-O-4 linkage has been proposed both to primarily undergo homolytic bond cleavage routes,[119–123] as well as concerted cleavage, primarily *via* retro-ene fragmentation.[124–128] Both for the numerous studies on β-O-4 bond cleavage, and generally in lignin model compound efforts, the model compounds, temperature, pressure, experimental equipment, and theoretical methods to probe these mechanistic questions often differ between studies and laboratories. As such definitive conclusions to date about how β-O-4 bonds are cleaved thermally is challenging and likely biased by the methods being employed. Emerging computational methods to build lignin libraries[129,130] and simulate the resulting chemistry[131] coupled to further advances in high resolution spectroscopy and continued development of theoretical techniques to probe the thermal breakdown of lignin will hopefully eventually shed light on the chemistry of lignin at high temperature.

4. Lignocellulose fractionation

Biomass fractionation technology is located at the heart of the biorefinery. It determines the fate of the individual lignocellulose constituents, and therefore, it can both widen or limit the array of downstream valorisation possibilities, especially regarding lignin (*i.e.* because of structural alteration, sulfur incorporation, *etc.*). Many fractionation approaches have been developed to date, ranging from traditional paper making to more sophisticated and environmentally friendly innovations. Each of these methods results in a specific lignin product, which can be isolated in the form of (i) a solid residue, (ii) a lignin precipitate, or even directly as (iii) a depolymerised product mixture.[132] The following section discusses the extensive and diverse domain of biomass fractionation, with particular attention to the associated lignin characteristics.

The various fractionation technologies are divided into two distinct classes. The first class covers methods that focus on the liberation of lignin from the biomass matrix (*i.e.* delignification), while the (hemi)cellulose carbohydrates are preserved in the form of a delignified pulp. These strategies are discussed in Section 4.1. Depending on the particular method, the lignin is isolated as a solid lignin precipitate (LP) or as a depolymerised lignin oil (DL). The second class of lignocellulose fractionation strategies comprises methods that target the conversion and solubilisation of the carbohydrate fractions (Section 4.2). Herein, lignin is mostly isolated in the form of an insoluble lignin residue (LR) or as a lignin precipitate (LP). Graphical overviews of each fractionation class are presented in Fig. 8 and 11, respectively, with indication of typical process conditions, lignin characteristics, product names, *etc.* The abbreviations LR, LP, and DL are used in these figures to denote the type of lignin product.

4.1 Fractionation based on delignification

4.1.1 Alkaline delignification methods. Alkaline media promote biomass delignification and lignin solubilisation (Section 3.1), and they are therefore frequently applied in the pulping industry. Kraft pulping is the main pulping process, producing over 90% of all chemical pulps.[8] Its global dominance is due to (i) the high quality of the resulting pulp, (ii) the integrated recovery of the pulping chemicals, and (iii) the self-sufficiency of the process in terms of energy demands.[8,21] During kraft pulping, wood is processed in an aqueous solution of NaOH and Na_2S, termed white liquor. The unique feature of this liquor is the presence of HS^--ions,[21,82,83,89] which improves the selectivity of pulping by enhancing delignification and lignin depolymerisation (Fig. 4), without concurrently accelerating carbohydrate solubilisation.[133] Nonetheless, the harsh alkaline environment induces severe lignin degradation and repolymerisation reactions (Section 3.1).[21,73] The spent processing liquor, which contains the solubilised lignin, termed black liquor, is mostly incinerated to recuperate energy and the pulping chemicals, hereby closing the mass and energy balances of the pulp mill. Alternatively, the solubilised lignin may also be isolated from the black liquor, for instance *via* precipitation induced by acidification.[48,134] This precipitate, termed kraft lignin, is highly condensed and contains a low amount of residual β-O-4 bonds.[73,75,135,136] Furthermore, kraft lignin also incorporates sulfur in the form of thiol groups,[73] which can complicate downstream valorisation (*e.g.* catalyst poisoning).[137]

Sulfite pulping is the second most important chemical pulping process, but its market share has decreased drastically ($<5\%$) with the rise of the more versatile and efficient kraft process.[48,76] Sulfite pulping can proceed both in alkaline, pH-neutral, or acidic conditions,[73,134,138] which is regulated by the choice of the (bi)sulfite salts. Irrespective of the pH, reactive α-positions are sulfonated, leading to benzyl sulfonate groups.[73,134,138] The presence of these sulfonate groups increases the water solubility, even at low pH.[73,76] The resulting lignosulfonate can be isolated,[76] for instance *via* ultrafiltration, extraction, or precipitation, and it is usually obtained as a salt (Na^+, NH_3^+, Mg^{2+}, Ca^{2+}).[48] Lignosulfonates are typically highly degraded (*i.e.* newly formed C–C linkages and decreased β-O-4 content),

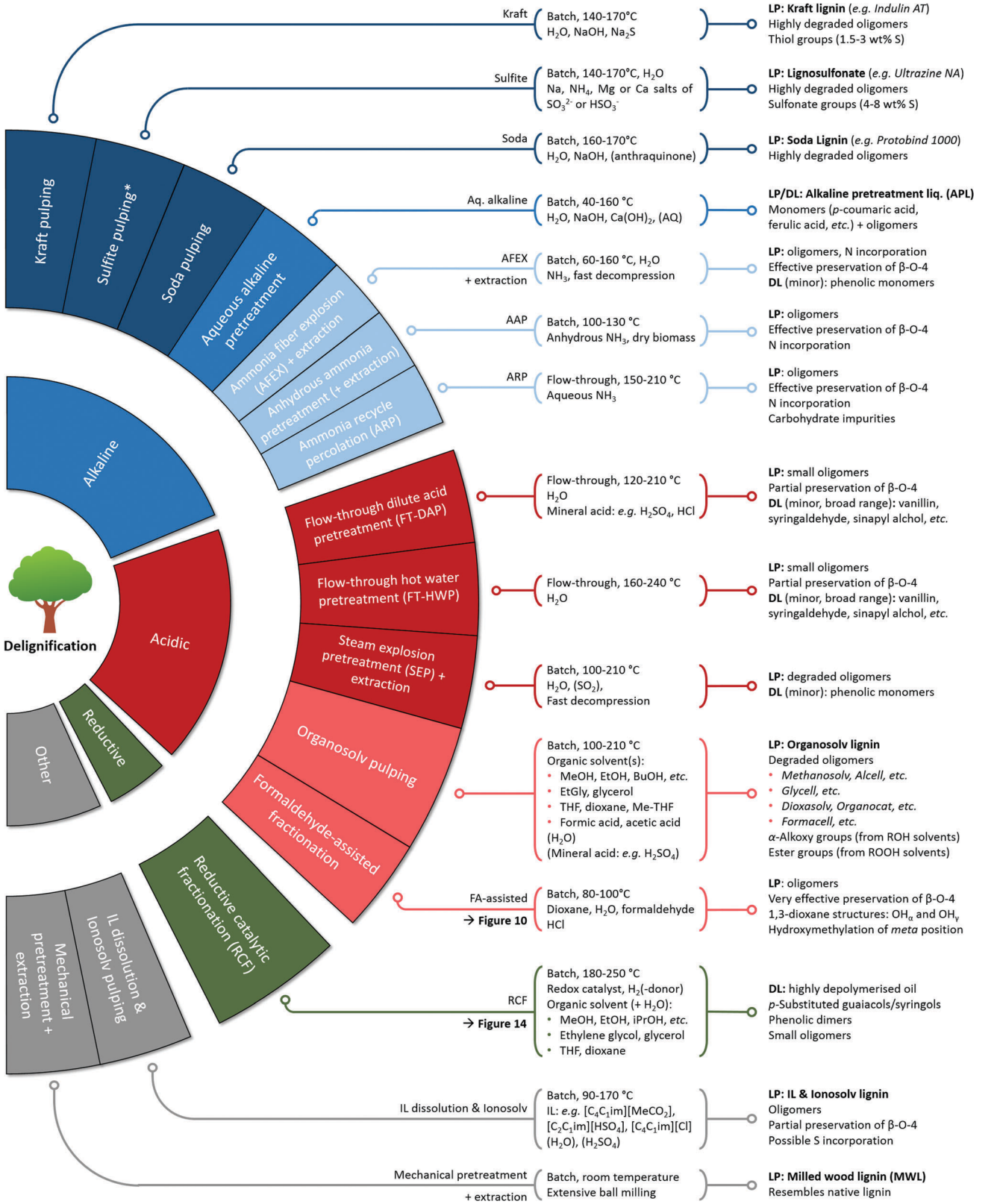

Fig. 8 Biomass fractionation methods, part I: methods focussing on the extraction of lignin (*i.e.* delignification of biomass). The extracted lignin can either be obtained as a solid lignin precipitate (LP) or as a depolymerised lignin oil (DL). The (holo)cellulose is retrieved in the form of a solid pulp. *Sulfite pulping can be performed under alkaline, pH-neutral or acidic conditions.

and have a higher sulfur content (4–8 wt%) compared to kraft lignin.[73,76,139,140]

The third traditional pulping process is soda pulping. It is related to Kraft pulping, with the major difference that soda pulping does not implement Na_2S.[21,73,134] Due to the absence of a strong nucleophile, alkaline depolymerisation (NaOH) proceeds less efficiently while competing reactions occur to a larger extent, as explained in Section 3.1 (Fig. 4). Soda pulping has historically been exploited to produce pulps from non-woody biomass (straw, miscanthus, flax, sugar cane, bagasse, *etc.*).[48,73,134] Non-woody biomass typically has a lower lignin content, a more open structure, and a larger portion of alkali-labile ester linkages. The efficiency of soda pulping can be increased by the addition of anthraquinone (AQ). It is suggested that AQ promotes reductive cleavage of ether bonds and simultaneously limits the degradation (peeling) of carbohydrates by operating as a redox shuttle (quinone–hydroquinone).[8,141,142] A major advantage of soda(-AQ) pulping is that a sulfur-free lignin is acquired, in contrast to kraft and sulfite pulping.[73] Soda(-AQ) lignin is typically characterised by a low β-O-4 content,[75,136,143] and can be isolated through precipitation.

Related to soda pulping is aqueous alkaline pretreatment (*e.g.* NaOH, $Ca(OH)_2$), which has mainly been studied on herbaceous biomass.[144–150] The major difference compared to soda pulping is the lower severity of the treatment. As demonstrated for aqueous NaOH pretreatment of corn stover, *circa* 55 wt% of the original lignin can be extracted into the liquor,[144] termed alkaline pretreatment liquor (APL).[144–147] Another 35 wt% can be removed by washing the residual solids with water.[144,151] The APL is rich in monomeric phenols such as *p*-coumaric acid, ferulic acid, and vanillic acid, which are obtained from hydrolysis of ester linkages.[144,145] These phenolic compounds can constitute up to 27 wt% of the original corn stover lignin.[144] In addition, the APL contains lignin oligomers,[144,145] as well as carbohydrate-derived hydroxy acids (*e.g.* lactic acid, glycolic acid) resulting from alkaline degradation of solubilised carbohydrates.[144] The aqueous wash phase only contains high molecular weight lignin oligomers.[151]

In addition to NaOH-based techniques, several other alkaline fractionation methods rely on (liquid) ammonia. Liquid ammonia is able to solubilise or redistribute lignin, while effectively preserving the carbohydrates.[152–156] An important advantage is the easy recovery of ammonia, owing to its high volatility. The most well-known technique in this category is ammonia fiber explosion/expansion (AFEX).[154,157–159] Herein, moist/wet biomass is reacted with ammonia under elevated pressure, which generates heat and induces both ammonolysis and hydrolysis of LCC and ester linkages, resulting in partial solubilisation of the lignin polymer.[154,158,160] Subsequently, ammonia is evaporated by a rapid and explosive pressure release, which opens up the biomass structure and redistributes lignin and hemicellulose in the pretreated solids.[154,160] It should be stressed that AFEX itself does not fractionate the biomass constituents. Nevertheless, it facilitates subsequent lignin extraction (*e.g.* with an organic or alkaline solution), which can remove up to 50–65% of the lignin from AFEX-pretreated corn stover.[154] The isolated lignin is mainly composed of oligomeric fragments, in which the β-O-4 bonds are rather well preserved.[143,161] It also contains a small amount of phenolic monomers, including aldehydes (vanillin, syringaldehyde), acids (vanillic acid, *p*-coumaric acid, *etc.*) and amides thereof.[158]

Closely related to AFEX is anhydrous ammonia pretreatment (AAP).[155,156,162] Liquid anhydrous ammonia is known to be an excellent cellulose swelling agent, as it is able to penetrate the cellulose fibres, including the crystalline domains. Herein, it can interfere with the natural hydrogen bond network, resulting in a cellulose–ammonia complex, which in turn leads to an altered crystalline structure upon controlled removal of ammonia (*e.g.* by evaporation).[155,156,162,163] It has been proven that the restructured cellulose (a C_{III} polymorph) is more susceptible towards enzymatic hydrolysis compared to the native polymorphic form (C_I).[164,165] It should be emphasised that the applied biomass for AAP should have a very low moisture content because water obstructs the formation of the C_{III} polymorph, which is one of the major differences with AFEX. A second dissimilarity is the absence of an explosive pressure release. Instead, retaining ammonia in the liquid state under high pressure enables to instantly extract the solubilised biomass constituents, primarily lignin. This specific operation of AAP is also termed extractive ammonia pretreatment (EAP).[155] EAP induces only minor structural lignin degradation, and was recently shown to enable 44% lignin extraction from corn stover.[155,162] An alternative operation encompasses (controlled) evaporation of ammonia after AAP, followed by mild extraction of the lignin with an aqueous NaOH solution (*e.g.* 0.1 M at 25 °C). This variant allows for higher lignin quantities to be extracted (up to 65% using corn stover), and tends to be more practical with respect to NH_3 recovery and lignin extraction. In both cases, β-O-4 bonds remain intact and nitrogen gets incorporated in the form of hydroxycinnamoyl amides (*i.e.* coumaroyl amide and feruloyl amide).[156,162]

A third ammonia-based delignification technique is ammonia recycle(d) percolation (ARP). In this flow-through process, lignin is continuously extracted by an aqueous ammonia solution,[159,166–171] which enables high degrees of delignification (up to 85% for corn stover).[159,166] Additionally, also a substantial fraction of the hemicellulose (*e.g.* 50–60%)[166] is extracted from the biomass. The solubilised lignin can be precipitated from the extraction liquor by evaporation (and recycling) of ammonia.[168] The resulting precipitate can contain a considerable amount of carbohydrate impurities (up to 20 wt%),[143] which can be removed almost entirely *via* a mild acid-catalysed hydrolysis procedure, without compromising the lignin structural integrity.[171] Bouxin *et al.* demonstrated that β-O-4 linkages could be well preserved during ammonia percolation of poplar wood,[135,171] although it should be mentioned that in this particular study the isolated lignin yield and delignification degree were rather low, 31% and 58% respectively (importance of lignin yield: see Section 4.3.1).[171] As with all ammonia-based fractionation methods, a small amount of nitrogen is incorporated as well (1–2 wt%).[143,171]

4.1.2 Acidic delignification methods. Dilute acid pretreatment (DAP) primarily intends to decrease the hemicellulose content

of the biomass, but additionally affects the lignin (Fig. 5).[90,91] Lignin fragments released upon DAP are partially soluble in acidic hot water, but subsequently condense and redeposit on the biomass surface if operated in batch mode.[172–175] As a result, the lignin structure is altered, while the lignin content is not effectively decreased by batch-DAP. Contrarily, when operating in flow-through (FT) mode, the solubilised lignin fragments are removed from the heating zone, which limits the extent of structural alteration and redeposition.[172–174,176] Hence, FT-DAP can be considered as an effective method to extract lignin (and hemicellulose). FT-DAP removes substantially more lignin than the analogues process in batch mode.[172–174,176] The hydrolysate contains hemicellulose carbohydrates (*i.e.* monomers and oligomers), lignin oligomers, and a smaller fraction of lignin monomers. It is expected that β-O-4 bonds in the oligomeric fraction are partly preserved, based on indirect information from related studies.[161,177–181] Complete isolation of the extracted lignin from the hydrolysate is difficult because precipitation is not effective for low molecular weight, oxygenated compounds.

A similar approach is FT-hot water pretreatment (FT-HWP),[173,174,176,182] which can be regarded as an autocatalytic form of FT-DAP, and is therefore also termed autohydrolysis. The acidity during FT-HWP is provided by (i) the increased dissociation of water at elevated temperatures,[182] and (ii) released organic acids from the biomass (*e.g.* acetic acid from acetate groups).[90,91,183] During FT-HWP, lignin undergoes acidolysis and acid-catalysed condensation, albeit to a lesser extent compared to FT-DAP.[90] The majority of the extracted lignin constitutes oligomeric fragments,[184] and it is assumed that β-O-4 bonds are at least partly preserved.[177–181] A small amount of monomeric phenols is obtained, comprising a broad range of compounds (up to 30), including *p*-hydroxybenzoic acid, vanillin, syringaldehyde, and sinapyl alcohol.[184–186] It has been postulated that vanillin, syringaldehyde, and their respective acids arise from oxidative degradation (*i.e.* oxidative cleavage of C_α–C_β bonds).[184,186,187] As with FT-DAP, isolating the extracted lignin *via* precipitation is difficult.

An alternative acidic method is steam explosion pretreatment (SEP),[91,188–194] which combines features of both AFEX and HWP/DAP. A treatment with pressurised steam/water (autohydrolysis) is followed by an explosive pressure release that opens up the lignocellulose matrix and physically disrupts the ordered fibrous structure.[192,193] Although SEP itself does not effectuate substantial biomass delignification,[91] it facilitates subsequent lignin extraction with an organic or alkaline solution (in analogy to AFEX).[188–190,192] The lignin can be separated from the co-extracted hemicellulose-derived products by precipitation (through acidification, water addition or evaporation of the organic solvent). Depending on the process severity, the lignin undergoes moderate to severe acid-catalysed degradation (50–100% loss of β-O-4).[66,91,191,192,195]

Whereas the above acidic delignification strategies all apply pure aqueous media, the degree of delignification can be significantly augmented by implementation of an organic solvent. This is the rationale behind organosolv pulping, a process wherein biomass is treated with an organic solvent, often in combination with mineral acids and/or water.[77,196–198] Due to the increased lignin solubility in the organic medium, lignin is more effectively extracted compared to (batch-)DAP/HWP. After the pulping process, lignin can be separated from the co-extracted hemicellulose fraction by precipitation from the pulping liquor, yielding organosolv lignin. Organosolv pulping thus enables the efficient fractionation of lignocellulose in its three major constituents: a solid cellulose pulp, a lignin precipitate, and an aqueous hemicellulose-derived stream.[197] Various solvents can be applied, including alcohols (methanol, ethanol, butanol),[102,199–206] polyols (ethylene glycol, glycerol),[207–210] cyclic ethers (THF, dioxane),[211–213] organic acids (formic acid, acetic acid),[198,214–216] and ketones (acetone, MIBK).[217–220] A popular choice are low boiling alcohols owing to their ease of recovery and low cost.[197] Organosolv pulping can take place both in presence or absence of an acid catalyst (autocatalytic fractionation).[197] In either case, lignin undergoes acid-catalysed depolymerisation and condensation (Fig. 5),[221] resulting in oligomeric fragments. The extent of structural alteration strongly depends on the process severity (Section 4.3.1).[205] Technical organosolv lignins obtained from industrially relevant (harsh) organosolv processes, such as the Alcell process (pulping with aqueous ethanol at *ca.* 195 °C),[222] contain only a limited amount of residual β-O-4 motifs.[75,205] Though, under certain conditions, lignin can be extracted with good preservation of β-O-4 linkages (*e.g.* dioxasolv pulping,[223,224] butanosolv pulping[204,205]). A signature feature of organosolv pulping with concentrated alcohols is the incorporation of solvent-derived alkoxy groups at the α-position of lignin's alkyl chains (Fig. 9).[64,204,205] It has been postulated that alkoxylation protects the β-O-4 structure from undergoing degradation and condensation reactions.[204,205] On the other hand, ester groups (formyl, acetyl) are incorporated upon employing carboxylic acids as pulping solvent (*viz.* esterification).[216,225]

A recent advancement in biomass delignification, demonstrated by Luterbacher *et al.*, encompasses the solvolytic extraction of lignin by using a dioxane/water mixture containing HCl and formaldehyde.[226] The innovative feature of this mild (80–100 °C) fractionation approach is the ability of formaldehyde to chemically stabilise lignin, hereby preventing acid-catalysed depolymerisation and repolymerisation reactions. Formaldehyde forms relatively stable 1,3-dioxane rings (*i.e.* acetal formation) by reacting with the OH_α- and OH_γ-groups of the alkyl side-chains in β-O-4 motifs (Fig. 10), which inhibits the formation of reactive carbenium ions (Fig. 5). As a result, an oligomeric lignin precipitate can be obtained, with almost complete preservation of β-O-4 bonds. Additionally, formaldehyde also partially blocks the electron-rich *meta* positions

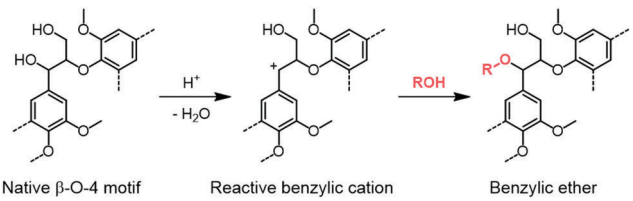

Fig. 9 α-Alkoxylation during acid-catalysed organosolv fractionation in concentrated alcohols.

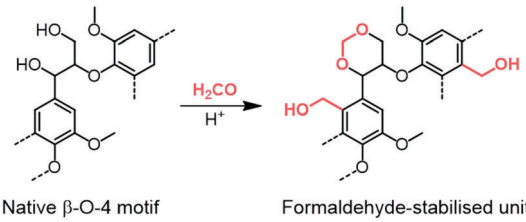

Fig. 10 Mechanism of lignin stabilisation during formaldehyde-assisted lignin extraction, forming acid-stable dioxane structures and *m*-hydroxymethyl groups.[226]

through the formation of *m*-hydroxymethyl groups (relative to the phenolic position).[226–230] Noteworthy, the subsequent formation of methylene cross-linkages such as in phenol formaldehyde resins does not occur, thanks to the optimised processing conditions.[229,230] A side-reaction of the formaldehyde-assisted delignification process is the incorporation of formaldehyde into the residual carbohydrate pulp. However, the grafted formaldehyde can be removed *via* an additional acidic hydrolysis step.[226]

4.1.3 Reductive catalytic fractionation. Reductive catalytic fractionation (RCF) combines solvolytic extraction of lignin with simultaneous reductive catalytic depolymerisation by a heterogeneous redox catalyst.[64,231–235] Hence, the process is closely related to organosolv pulping, but differentiates itself by producing a highly depolymerised lignin oil instead of a high molecular weight precipitate. The extent of delignification is mainly determined by the solvent type, reaction time, and temperature.[236] As in organosolv fractionation, low boiling alcohols (*e.g.* methanol, ethanol, isopropanol) are the most commonly used solvents, and are often mixed with water.[232,234,236,237] Furthermore, addition of an acidic co-catalyst (*e.g.* H_3PO_4, metal triflates)[238–242] can enhance the extraction of lignin as well as the removal of hemicellulose. The subsequent lignin depolymerisation (hydrogenolysis) and reductive stabilisation are governed by the redox catalyst. To this end, supported noble metals as well as Ni have been applied.[234,235,239,243,244] Since RCF combines fractionation with lignin depolymerisation, details on the obtained lignin products (substituted methoxyphenols) are discussed more elaborately in Section 5.

It should be noted that the carbohydrates are obtained as a (holo)cellulose pulp, together with the spent catalyst. Separation of the catalyst from the pulp is of utmost importance to guarantee the viability of the RCF process, but remains nevertheless difficult. Within this respect, the use of ferromagnetic catalysts[234,245] or a catalyst basket[233,366] has been demonstrated to enable a facile catalyst-pulp separation.

4.1.4 Other delignification techniques. Ionic liquid dissolution and ionosolv pulping are two interrelated fractionation approaches based on the action of ionic liquids (ILs). Depending on the applied IL, the entire lignocellulose substrate can be dissolved (IL dissolution) or the lignin and hemicellulose can be extracted (ionosolv pulping).[246] In the former process, cellulose can be precipitated from the product mixture by addition of an anti-solvent (organic or aqueous–organic solution),[246–251] prior to lignin precipitation in a subsequent step. A major advantage of this process is the potential to decrystallise cellulose,[247] which can facilitate downstream conversion. During ionosolv pulping, the second IL-based process, only lignin and hemicellulose are solubilised while the cellulose fraction remains in the form of a solid pulp. Hence, ionosolv pulping shows a strong resemblance to organosolv pulping, but can generally be performed at lower temperatures (<160 °C). The solubilised lignin can be precipitated from the pulping liquor.[246,252–256] For both IL-based fractionation techniques, partial β-O-4 cleavage occurs followed by repolymerisation, depending on the process severity.[135,161,251–253,256,257] It has been postulated that the extent of β-O-4 cleavage strongly depends on the type of anion (*e.g.* sulfate, acetate, phosphate), which can act as nucleophile. The cation on the other hand has only a minor contribution.[246,258] Furthermore, sulfur can be incorporated upon applying sulfur containing anions (*e.g.* sulfate, sulfonate, sulfamate).[246,258] Other considerations, which are inherently connected to the use of ionic liquids, include IL cost, toxicity and recuperation.[246,250,253,259]

Mechanical pretreatment can also facilitate biomass fractionation. This principle forms the basis for the isolation of milled wood lignin (MWL).[260–263] MWL is obtained through extensive ball milling of wood at room temperature, followed by lignin extraction with an organic solution like dioxane/water. In this way, a lignin substrate can be obtained that is structurally similar to native lignin.[260–263] Despite the high content of β-O-4 bonds, MWL isolation is however not relevant on an industrial scale. Long milling times (days–weeks) are required and the degree of delignification is generally low (<35%).[67] The method is only of interest for analytical and experimental purposes because it provides a native lignin surrogate.

Finally, oxidative delignification, although usually applied in the context of pulp bleaching, can also be used to remove lignin from raw lignocellulosic biomass. This can either be performed under acidic (*e.g.* in acetic acid or peracetic acid)[264] or alkaline conditions (*e.g.* in aqueous NaOH[265–267] or lime[264]) and usually with hydrogen peroxide[265–267] or oxygen[264] as oxidant. Herein, lignin is transformed through oxidative (next to acid- and base-catalysed) pathways, resulting in a mixture low-molecular weight products which likely comprise phenolic, quinone, and ring-opened (aliphatic carboxylic acid) structures (Section 3.4).[84,108,109] Since only very little information is available about the structure of the extracted lignin, oxidative delignification is not included in Fig. 8.

4.2 Fractionation based on carbohydrate conversion

The second category of fractionation methods, summarised by Fig. 11, encompasses lignocellulose fractionation approaches that are based on the conversion (hydrolysis or thermolysis) of carbohydrates. A lignin (side-)product can be recovered either in the form of an insoluble lignin residue (LR), or as a lignin precipitate (LP). In analogy to Fig. 8, these abbreviations are also used in Fig. 11 to indicate the different types of lignin.

4.2.1 Acid-catalysed carbohydrate conversion. A traditional approach to convert lignocellulosic carbohydrates into monosaccharides is through concentrated acid hydrolysis (CAH).[268]

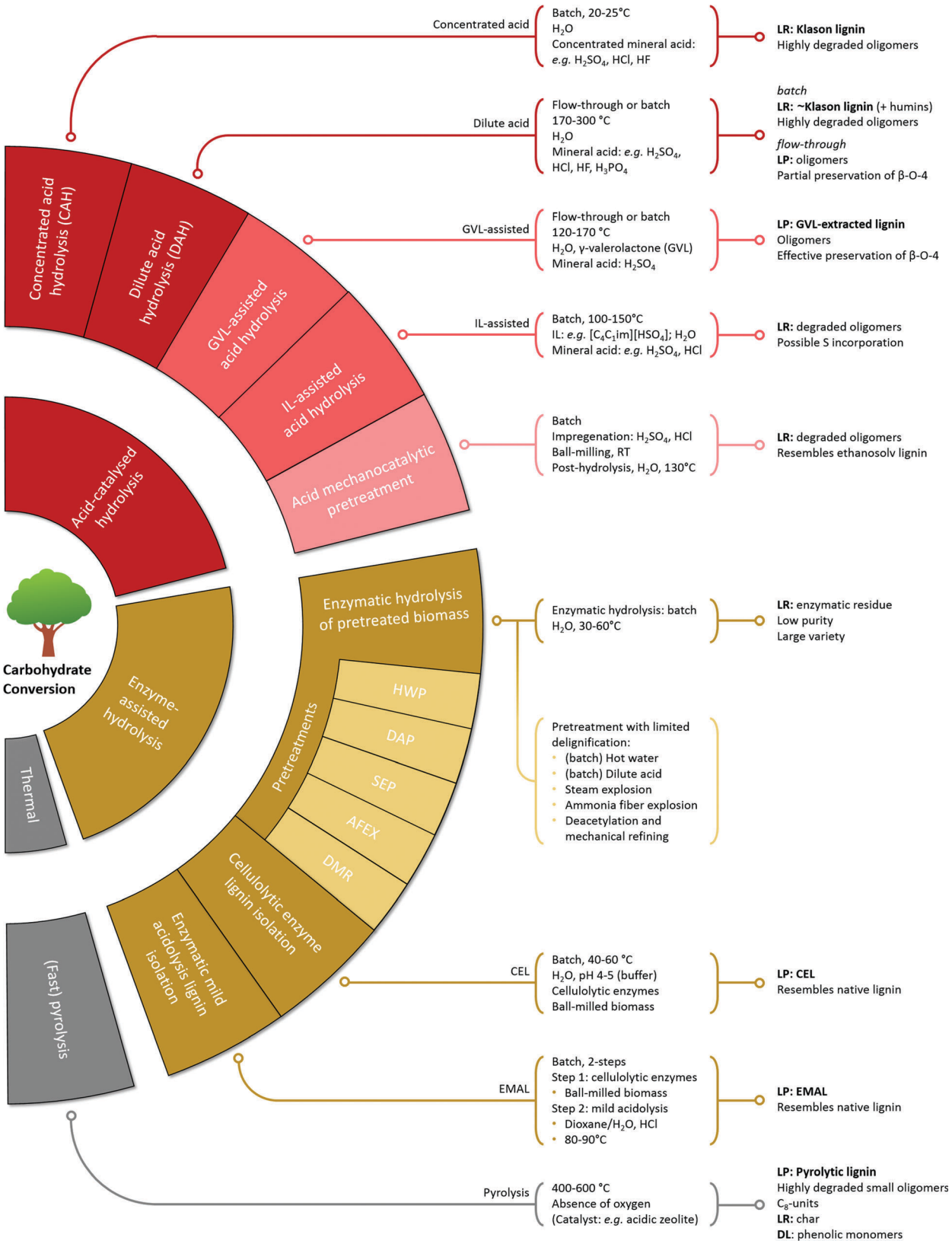

Fig. 11 Biomass fractionation methods, part II: methods focussing on the deconstruction of the carbohydrate polymers. The lignin fraction can either be obtained as a solid lignin residue (LR) or as a lignin precipitate (LP).

In this method, concentrated mineral acids (*e.g.* 72 wt% H_2SO_4) are applied to digest raw biomass at room temperature, resulting in an aqueous solution of mainly carbohydrate oligomers. A post hydrolysis step with dilute acid (0.5–5 wt%) at elevated temperature (*e.g.* 100 °C) is required to yield sugar monomers (80–100% yield).[268–270] The lignin fraction undergoes severe degradation by acid-catalysed cleavage of ether bonds and repolymerisation (Section 3.2; Fig. 5). As a result, the majority of the lignin is obtained as a highly condensed insoluble residue, named Klason lignin. The amount of Klason lignin can be determined gravimetrically, and is a standard analytical method to measure the (acid-insoluble) lignin content of lignocellulosic biomass.[271,272] Apart from the Klason lignin, a minor fraction is present as acid soluble lignin (ASL), which can be determined spectrophotometrically. ASL is suggested to comprise oxygenated lignin monomers and oligomers. Major drawbacks of concentrated acid hydrolysis are corrosion issues and the difficulty of recovering and regenerating the mineral acid.

Related to CAH is dilute acid hydrolysis (DAH).[268,273,274] The lower acid concentration (<5 wt%) is compensated by operating at a higher reaction temperature (>170 °C).[268] In this harsh environment, both cellulose and hemicellulose are hydrolysed. The lignin fraction undergoes acid-catalysed degradation, and is recovered as a highly altered insoluble residue if operated in batch mode. Furthermore, the residue can contain humins (also termed *pseudo*-lignin), which arise from acid-catalysed degradation of carbohydrates.[275–278] Lignin and carbohydrate degradation can be reduced by operating in flow-through mode (FT-DAH), which enables a shorter residence time of solubilised, reactive species. This alternative operation mode yields less degraded lignin, similar to lignin obtained from FT-DAP/HWP. The majority of this lignin fraction comprises oligomers which can be precipitated from the hydrolysate. A smaller portion consists of monomeric phenols such as vanillin, syringaldehyde, coniferyl alcohol, *etc.*[178–180,187] The oligomeric fraction is reactive towards depolymerisation, which indicates that a significant part of the β-O-4 bonds are preserved.[178–180]

A recent innovation in the field of acid-catalysed hydrolysis, demonstrated by Dumesic *et al.*, involves the use of γ-valerolactone (GVL)/water mixtures in combination with H_2SO_4. GVL facilitates the complete solubilisation of the biomass, by promoting both (hemi)cellulose deconstruction and lignin solubilisation.[279] The carbohydrates can be acquired as mono- and oligosaccharides (70–90% yield)[279] or as secondary products such as levulinic acid and furfural,[280] whereas the solubilised lignin can be precipitated through addition of water to the mixture.[280,281] Alternatively, the lignin precipitate can also be recovered by CO_2 extraction of the GVL/water mixture, circumventing the need for additional water.[162,172] Hence, by applying CO_2-based separations, high concentrations of monosaccharides in the aqueous phase can be obtained (up to 127 g L^{-1}).[279] With respect to the lignin fraction, GVL-based fractionation enables mild processing conditions, which favours the preservation of β-O-4 ether bonds.[281]

Acid hydrolysis of lignocellulose can also be performed in acidified ionic liquids.[246,282–285] The use of ionic liquids enables the solubilisation of the lignocellulosic biopolymers (*vide supra*, Section 4.1.4), making the glyosidic bonds more accessible towards hydrolysis. Hence, carbohydrate hydrolysis can occur more efficiently in ILs compared to aqueous systems.[284] Lignin undergoes acid-catalysed degradation reactions (Fig. 5) leading to a residue with low content of β-O-4 ether bonds.[282–284] Sulfonate groups from the ILs can be incorporated as well.[283]

Finally, another alternative acid-catalysed fractionation method is mechanocatalytic depolymerisation, which has been extensively studied by Rinaldi *et al.*[286–292] This technique relies on milling of acid-impregnated biomass, hereby fully converting the substrate into water-soluble products (oligosaccharides and lignin fragments). Upon applying a post-hydrolysis step, high yields of monosaccharides can be obtained.[286–289] Simultaneously, a lignin precipitate is formed during the acid-catalysed saccharification,[287,288] which closely resembles ethanosolv (50/50 ethanol/water, 180 °C) lignin in terms of β-O-4 content.[287] The obtained precipitate comprises oligomers characterised by a rather low amount of β-O-4 ether bonds.[287,290,292] It has been suggested that most of the structural alteration (depolymerisation and condensation) occurs during the actual mechanocatalytic depolymerisation rather than during the post-hydrolysis step.[287] Nevertheless, part of the repolymerisation can be avoided by performing the post-hydrolysis step in a biphasic system comprising water/MeTHF, thereby yielding a lignin polymer with lower molecular weight compared to the lignin precipitate obtained from monophasic aqueous saccharification.[291] It was postulated that the extraction of lignin fragments in the MeTHF phase protects them from recondensation.[291]

4.2.2 Enzymatic-assisted carbohydrate conversion. Enzymatic hydrolysis is a common strategy to liberate monosaccharides from the carbohydrate polymers residing in lignocellulosic biomass. Concurrently, a solid residue is obtained which contains the water-insoluble lignin-rich fraction, together with residual carbohydrates. Because many physico-chemical factors of raw biomass hinder the direct biological deconstruction of (hemi)cellulose, a pretreatment step is usually applied to reduce biomass recalcitrance. In case the pretreatment step does not effectuate substantial delignification, a similar lignin-enriched residue is obtained upon enzymatic hydrolysis of the pretreated biomass.[293–296] The most common pretreatment methods that do not induce extensive delignification are HWP,[91,297,298] DAP,[91,299,300] SEP,[91,191–193,301] AFEX,[91,154,160] and deacetylation and mechanical refining (DMR).[302–304] Other, less conventional, pretreatment strategies include plasma pretreatment[305] and ultrasound pretreatment.[306,307] The residues obtained from enzymatic hydrolysis have a low lignin purity (*e.g.* ashes, residual carbohydrates, protein, *etc.*),[293–295,301,308] but receive increasing attention to be used as lignin resource for further valorisation.[293,294,309,310] The purity as well as the extent of structural alteration strongly depend on the type and severity of the pretreatment method.

Apart from the industrially relevant residues discussed above, two enzymatic laboratory scale procedures exist to isolate a pure lignin with minimal structural alteration and high β-O-4 content. Both methods rely on extended enzymatic hydrolysis (typically for 48 h) of ball milled wood by cellulolytic enzymes.[67]

Extraction of lignin from the residual solids (*e.g.* with dioxane/water) followed by precipitation yields cellulolytic enzyme lignin (CEL).[262,311–314] The isolated yield is generally low (<35%), but the β-O-4 linkages are well preserved.[67] Alternatively, following the enzymatic hydrolysis, the insoluble material can be submitted to an additional mild acid hydrolysis step (HCl in dioxane/water), to cleave LCC linkages and solubilise lignin.[67,315] Subsequent precipitation yields enzymatic mild acidolysis lignin (EMAL), which is obtained in significantly higher yields (25–65%) compared to CEL.[67,315–318] Nevertheless, the isolation of EMAL is not applicable on an industrial scale, as is also the case for the isolation of CEL and MWL. These isolated lignins are only relevant in a research context, for instance to study the depolymerisation of (close-to-)native lignin.

4.2.3 Thermal carbohydrate conversion: pyrolysis. (Fast) pyrolysis of lignocellulose is an extensively investigated process wherein biomass is decomposed thermally (400–600 °C) in absence of oxygen, resulting in gaseous products and char.[319,320] The generated char is mainly derived from lignin.[320] Subsequent condensation of the gases yields a liquid product, and is referred to as pyrolysis oil or bio-oil. In order to maximise the oil yield, high heating rates (300–1000 °C min^{-1}) and short residence times (1–2 s) are preferred.[319] Although oil yields up to 75 wt% can be reached, the obtained bio-oil is unstable, has a low energy content, a high water content, and is immiscible with petroleum-based fuels.[319] *In situ* or *ex situ* catalytic upgrading is therefore required to produce a fuel-compatible liquid. The interested reader is referred to dedicated literature on this topic.[71,319–323]

The pyrolysis bio-oil fraction contains both Carbohydrate- and lignin-derived products,[71] with the latter including phenolic monomers and oligomers. The thermally formed lignin monomers can be present in considerable amounts (up to 20 wt% of initial lignin), and comprise a wide array of compounds (phenols, catechols, guaiacols *etc.*).[71,324–328] The lignin-derived products are more hydrophobic compared to the carbohydrate-derived compounds (furfural, anhydrosugars, short aldehydes and acids, *etc.*),[320] which makes it possible to precipitate most of the lignin fraction from the bio-oil, for example by addition of water. The phenolic monomers are however expected to remain mostly in the liquid phase. The as-formed precipitate is termed pyrolytic lignin, and is highly condensed and degraded.[329–332] It is composed of short oligomers (DP 4–9),[329] which are mainly built from C_8-rather than (native) C_9-units due to degradation of the C_3-side-chain.[24,66]

4.3 Critical discussion on fractionation

4.3.1 Structural analysis and comparison of isolated lignins. Lignocellulose fractionation methods induce various alterations in the native lignin structure, depending on the method employed and the process severity. These structural changes affect the reactivity towards depolymerisation of the isolated lignin, which is mostly assessed by measuring the β-O-4 content. The β-O-4 content is one of the most important lignin characteristics,[135,143,205,333] though, it is not the sole factor that governs reactivity. Other structural features such as the OH-group content, molecular weight, and the presence of impurities might exert an effect as well.

Measuring the β-O-4 content is most often performed *via* 2D ^{1}H–^{13}C heteronuclear single quantum coherence nuclear magnetic resonance spectroscopy, abbreviated as HSQC NMR.[334] Although this method delivers valuable structural information, performing quantitative analyses is difficult.[75,256] Absolute values should therefore be interpreted with caution and they are most valuable in a comparative context. Destructive analytical methods like thioacidolysis[335,336] and nitrobenzene oxidation[337,338] on the other hand provide an alternative quantitative measure of lignin reactivity: a higher yield of volatile (monomer) products is acquired for more reactive lignins (having a higher β-O-4 content). These tools can be regarded as analytical depolymerisation methods, complementary to HSQC NMR.

Owing to variations in analytical protocols, it is often difficult to unambiguously compare characterisation studies on lignin. This issue is overcome by comparative studies that systematically apply a well-defined set of analytical methods on different isolated lignins. For example, Bouxin *et al.* characterised four different types of lignin (wheat straw soda, poplar organosolv, wheat straw AFEX, and poplar ARP lignin) with HSQC NMR and thioacidolysis.[143] Although different absolute values were obtained with both techniques, a clear trend could be deduced. Both techniques demonstrated that the β-O-4 bonds in poplar ARP and wheat straw AFEX lignin were well preserved (HSQC NMR: 45% and 37% of interunit linkages), in contrast to poplar ethanosolv lignin (12%) and wheat straw soda lignin (4%).[143] In a follow-up study, Bugg *et al.* characterised a set of seven different lignins.[135] Based on HSQC measurements, they concluded that poplar ARP lignin contained the highest proportion of β-O-4 bonds (48%), followed by oak dioxasolv lignin (40%), eucalyptus ethanosolv lignin (16%), miscanthus ionosolv lignin (10%), and eucalyptus kraft lignin (<1% β-O-4).

It should be noted that the lignin reactivity also depends on the biomass type and the fraction of native β-O-4 bonds. Large structural and compositional variation exist between hardwoods, softwoods and herbaceous feedstocks, and between species within these categories (Section 2.1).[47,66,74] As can be intuitively understood, these difference are also reflected by the respective isolated lignin, and can furthermore influence the efficiency of fractionation. Hence, it is difficult to unambiguously compare fractionation methods if diverse biomass sources are used. Nonetheless, in the study by Bugg *et al.*, a large difference in β-O-4 bond content was measured for two types of hardwood organosolv lignin, *viz.* oak dioxasolv (40%) and eucalyptus ethanosolv lignin (16%). This vast dissimilarity illustrates that the structural characteristics of lignins are strongly affected by the specific process conditions.

In a recent comparative study by Barta *et al.*, a series of 22 organosolv lignins were characterised, including self-synthesised as well as commercial (technical) lignins.[205] The β-O-4 content of the organosolv lignins varied strongly, ranging from 2 to 62% as measured by HSQC NMR.[205] Hence, the fractionation process and severity clearly have a drastic impact on the lignin reactivity. In general, increasing the process severity, for instance by increasing the temperature, time, or

acidity, increases the degree of repolymerisation and decreases the lignin reactivity. On the other hand, a more severe process in general allows to remove more lignin from the substrate (*i.e.* delignification), and recover more lignin as a precipitate (*i.e.* isolated lignin yield). Therefore, the delignification, isolated lignin yield, and the lignin reactivity are closely intertwined, with a high isolation yield usually concurring with low reactivity towards depolymerisation, and *vice versa*. For example, in the study by Barta *et al.*, the high-yield commercial/technical organosolv lignins displayed a significantly lower β-O-4 content (8% β-O-4 on average) compared to low-yield self-synthesised lignins (39% β-O-4 on average).[205,339] This points out the importance of indicating the isolated lignin yield when assessing the lignin reactivity towards depolymerisation. Similar as comparing product selectivities at similar reagent conversions, the lignin reactivity is ideally evaluated at similar isolated lignin yields. Unfortunately, this aspect is often neglected in studies on the structural analysis of isolated lignins.

In a recent study, Bruijnincx *et al.* thoroughly characterised a set of six (high-yield) technical lignins which are frequently used in depolymerisation studies, including softwood kraft lignin (Indulin AT), grass/wheat straw soda lignin (Protobind 1000), hardwood ethanosolv lignin (Alcell) and various other organosolv lignins.[75] They concluded that all types, including the organosolv lignins, were strongly degraded by the isolation method, as expressed by the low fraction of inter-unit ether bonds (3–10% as measured by HSQC NMR). Noticeably, the studied poplar organosolv lignin showed a much lower β-O-4 content than the poplar organosolv lignin analysed by Bouxin *et al.* (3 *vs.* 12%).[77,90] Furthermore, Bruijnincx *et al.* determined a similar β-O-4 content for softwood kraft lignin as for the organosolv lignins, which is in sharp contrast to observations made by Bugg *et al.*[77,101] These differences may be explained by (i) variations in analytical procedures and/or (ii) by variations in the process severity, which again underpins the importance of indicating the isolated lignin yield.

In summary, the above comparative studies share some common observations, but on the other hand highlight that making a generalised comparison between isolated lignins is extremely difficult (see also Section 5.3.2). An overwhelming amount of variables come into play, both with respect to the fractionation method itself as well as to the actual analysis. Moreover, no biomass sample is the same, not even two samples from the same species.[340,341] Hence, the fractionation methods outlined in Fig. 8 and 11 do not concur with one specific lignin structure. Instead, each lignin has its own unique chemical properties, determined by (i) the fractionation method, (ii) the fractionation severity, and (iii) the biomass source. These aspects are outlined in Fig. 12. Ideally, to study the impact of a certain aspect on the lignin reactivity towards depolymerisation (*e.g.* fractionation method), the other parameters should be kept constant (*e.g.* biomass type and isolated lignin yield).

4.3.2 Preventing structural lignin degradation during fractionation. The development of industrially feasible lignin isolation methods that induce minimal structural degradation while achieving high lignin yield and purity, is a grand challenge in the lignin research field.[342] Notwithstanding that significant progress has been made, every isolation method results in structural alteration to a certain extent, which can negatively affect subsequent lignin conversion to chemicals (Section 5.3.2). As learned from the above literature survey, the quality of the isolated lignins can be augmented by taking into account at least one of the following two principles.

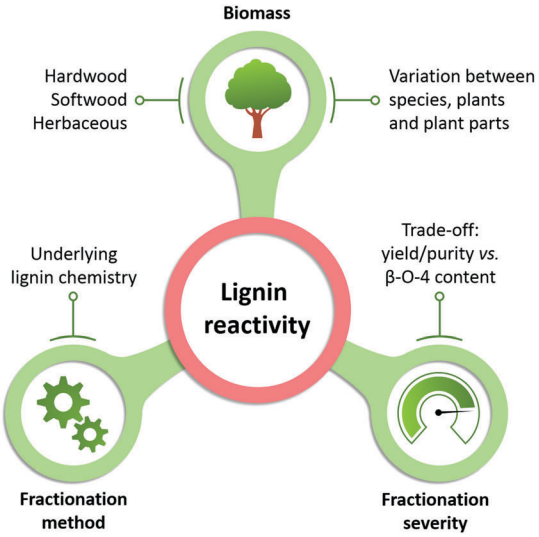

Fig. 12 The reactivity towards depolymerisation of any isolated lignin is determined by three elements: (i) the structural characteristics of the native lignin, (ii) the fractionation method and underlying mechanism, and (iii) the severity of the fractionation method, which is strongly linked to the isolated lignin yield/purity.

The first principle to minimise degradation is to keep the original lignin structure intact as much as possible, by preventing lignin depolymerisation. In other words, preserving the reactive β-O-4 bonds in lignin is one of the approaches to increase the potential for subsequent valorisation routes. Methods based on harsh acidic and alkaline conditions clearly do not meet this criterion. Classic pulping and carbohydrate hydrolysis methods evoke cleavage of β-O-4 ether bonds, hereby generating reactive intermediates that are prone to subsequent irreversible repolymerisation (Fig. 13). Milder processing conditions are more preferable to limit ether bond cleavage. For example, ammonia-based strategies (AFEX, AAP, ARP) tend to be milder compared to NaOH-based methods, and hence, preserve β-O-4 linkages more effectively. In addition, media that assist the solubilisation and/or saccharification of the biomass, for example GVL, offer a practical solution. In this way, the need for strong alkaline/acidic media or high temperatures is avoided, in contrast to purely aqueous-based methods. Finally, depolymerisation of β-O-4 motifs can be prevented *via* chemical stabilisation. For instance, the formation of stable 1,3-dioxanes through reaction with formaldehyde (Fig. 10) has been demonstrated as an effective β-O-4 preservation strategy.[226] α-Alkoxylation (Fig. 9) during acid-catalysed extraction of lignin with concentrated alcohols (*e.g.* *n*-butanol) has also been proclaimed to stabilise β-O-4 ether bonds.[204,205]

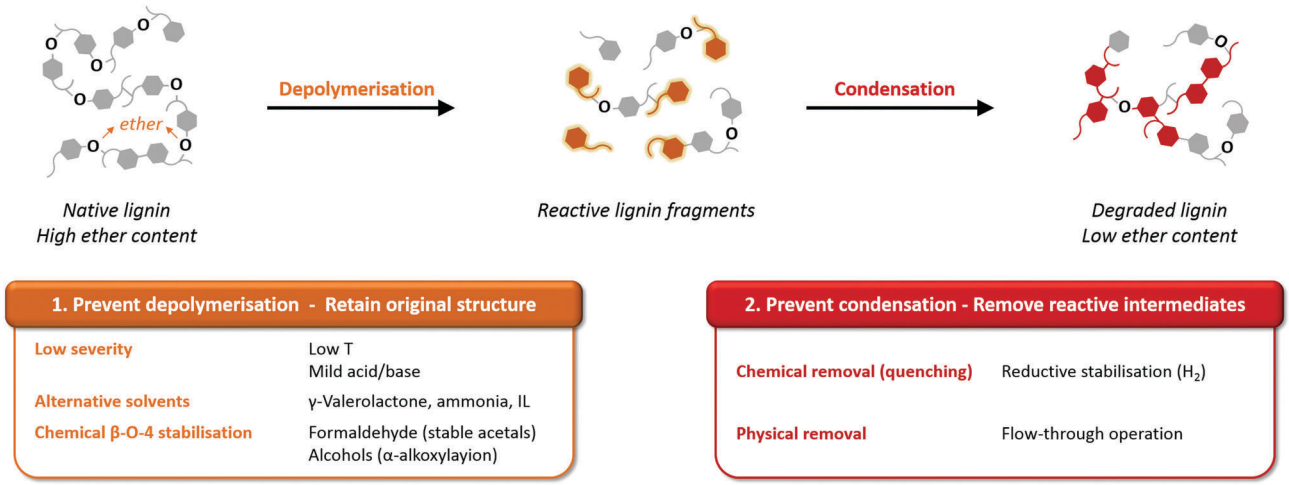

Fig. 13 Schematic illustration of lignin degradation (O-linkages represent ether bonds). Degradation can be prevented by (i) preventing depolymerisation and the formation of reactive intermediates, and/or by (ii) minimising subsequent repolymerisation reactions and the formation of new C–C bonds.

The second principle to prevent lignin degradation is to avoid repolymerisation of reactive intermediates. A practical method to inhibit the formation of stable carbon–carbon bonds is to physically remove these intermediates from the heating zone, by operating in flow-through mode instead of in batch operation. This methodology is applied during FT-DAP, FT-HWP, ARP, GVL-assisted processing and fast pyrolysis. However, flow-through operations often require a higher liquid-to-solid ratio (or gas-to-solid ratio) compared to their analogous batch processes,[343] consequently leading to more diluted product streams, a more energy intensive product work-up, and higher rate of solvent use, regeneration, and recycle. Alternatively, reactive intermediates can also be removed chemically, *i.e.* by quenching. This approach is for instance implemented during RCF. During this process, reactive intermediates are reduced (H_2 or H-donor) to stable end products. Interestingly, physical or chemical removal of reactive intermediates can also be applied in lignin depolymerisation processes (Section 5.3, Fig. 23).

Finally, on-purpose design of lignocellulosic biomass by means of genetic modification embodies a powerful complementary strategy to facilitate effective biomass fractionation/deconstruction. Pioneering work in this field by Chen and Dixon demonstrated that genetically reducing the lignin content in alfafla lines can increase the efficiency of DAP and enzymatic hydrolysis.[344] A noteworthy and more recent illustration encompasses the incorporation of labile ester linkages (*i.e.* monolignol ferulate conjugates; referred to as *zips*) in the backbone of poplar lignin, which augments the efficiency of delignification strategies.[345,346] Other plant engineering tactics to facilitate biomass deconstruction include the design of lignins with a lower polymerisation degree, less branching, augmented S/G ratio, less cross-linking with carbohydrates, and increased hydrophilicity.[51,62] Such *in vivo* alterations may enable fractionation under milder process conditions, hereby diminishing lignin depolymerisation (Principle 1) and/or repolymerisation (Principle 2). Notwithstanding, it should be mentioned that genetic modifications can also negatively impact plant growth (*e.g.* dwarfism).[347] Addressing these abnormalities is essential to exploit the full potential of bio-engineered crops, and requires deep understanding of the underlying regulatory pathways. For instance, Bonawitz *et al.* demonstrated that disrupting the transcriptional co-regulatory complex Mediator mitigates dwarfism in Arabidopsis C3′H mutants, with the engineered lignin almost exclusively containing H-units.[348]

4.3.3 Fractionation efficiency. Because this review focuses on lignin valorisation, the above discussion on biomass fractionation primarily sheds light on the obtained lignin (type, structure, and yield). Notwithstanding, it is important to note that fractionation techniques should also be evaluated in terms of fractionation efficiency, which can be interpreted as the yield and purity of the obtained fractions, *i.e.* lignin fraction and carbohydrate fraction(s). For example, the methods outlined in Fig. 8 ideally aim at fully extracting the lignin from the biomass, whether or not in combination with hemicellulose removal, while maximally preserving the (holo)cellulose portion in the residual solids. Hence, efficient biomass fractionation requires to find the right balance. It can be intuitively understood that this balance, just as the structural integrity of the lignin fraction, is determined by (i) the underlying fractionation chemistry, (ii) the fractionation severity, and (iii) the structural and compositional characteristics of the biomass (Fig. 12).

5. Lignin depolymerisation

As discussed in the previous section, a wide array of isolated lignins is available. It is commonly accepted that lignin is prone to structural degradation during biomass fractionation, resulting in a depletion of ether bonds and a concurrent increase in carbon–carbon bonds relative to the native lignin (Fig. 13). Since most depolymerisation methods are unable to cleave carbon–carbon bonds in lignin, an increase in carbon–carbon bond content (at the expense of cleavable ether bonds) lowers the potential for depolymerisation (or reactivity). As the theoretical

monomer yield from lignin is roughly proportional to the square of the relative content of cleavable inter-unit ether bonds,[21,244,349] a small decrease in ether bonds (and concomitant increase in carbon–carbon bonds) drastically lowers the monomer yield that can be achieved. To avoid this loss in reactivity, one suitable strategy is to perform lignin depolymerisation directly on native lignin residing in the lignocellulose substrate. In this way, biomass fractionation and lignin depolymerisation are executed simultaneously (see line 2 in Fig. 3). This intensified approach was briefly mentioned in the previous section on lignocellulose fractionation, and will be discussed in more detail in Section 5.1. Nonetheless, as explained in Section 4, traditional fractionation processes (kraft pulping, *etc.*) generate large volumes of degraded lignin streams, for which depolymerisation to valuable chemicals is an attractive valorisation opportunity. In addition, alternative fractionation processes are being developed that yield reactive isolated lignins with high β-O-4 content. (Principle 1 in Fig. 13). Therefore, depolymerisation of isolated lignins, either reactive or (partially) degraded, encompasses an important research field in the context of lignin valorisation and will be thoroughly reviewed in Section 5.2.

A difficulty in reviewing lignin depolymerisation is that studies generally report various quantitative measures, such as monomer yield, total product yield, oil yield, and lignin conversion. Because the latter three are defined in various ways,[350] this often does not allow for a straightforward comparison of lignin depolymerisation studies. For this reason, monomer yields are used as a measure of depolymerisation efficiency, and only studies that report monomer yields are discussed. Also, as this review focuses on the production of chemicals from lignin, the most relevant quantitative information is provided by the monomer yields. It should be stressed that the monomer yield is not the only conclusive factor, as selectivity is important as well. Therefore, to compare the various depolymerisation methods, both the monomer yield and the product selectivity in the monomer fraction are evaluated.

5.1 Depolymerisation of native lignin

In various lignocellulose fractionation methods, lignin depolymerisation can take place to a certain degree (see Section 4). While most of these methods don't specifically target extensive lignin depolymerisation, a few fractionation techniques are intentionally geared to yield a highly depolymerised lignin product. Umbrella terms such as 'lignin-first' or 'early-stage catalytic conversion of lignin' (ECCL) are often used to refer to (some of) these processes.[21,342] One particular process that combines lignin isolation and effective depolymerisation in one step, is RCF. This process, which was already briefly described in Section 4.1.3, has been extensively studied in recent years. RCF and related one-pot methods will be discussed in the next section. In Section 5.1.2, other methods that target native lignin depolymerisation will be addressed.

5.1.1 Reductive catalytic fractionation (RCF) and reductive one-pot methods. In RCF, which is also known as protolignin hydrogenolysis, native lignin is solvolytically extracted from the lignocellulosic biomass and simultaneously depolymerised in presence of a heterogeneous redox catalyst and a hydrogen source (H_2 or hydrogen donor).[64,195,226,231–245,342,351–366] A recent study has shown that lignin depolymerisation is mainly accomplished through solvolytic action (*e.g.* MeOH), while the catalyst's prior role is to reductively stabilise reactive intermediates, hereby largely avoiding repolymerisation.[366] As a result, the process yields a depolymerised lignin oil rich in phenolic monomers, dimers, and oligomers, next to a solid carbohydrate pulp which is amenable to further valorisation.[231,233,234,239,351] Hydrogen can be available as pressurised hydrogen gas, but can also originate from the solvent[234,235] or from lignocellulose itself.[232,352] The most common solvents are small alcohols (mainly methanol) and water/organic solvent mixtures such as water/dioxane and water/ethanol. The lignin product yield, delignification degree, and carbohydrate retention in the pulp strongly depend on the solvent,[235–237,353] additives,[238–242,244,245,354,355] and reaction temperature.[231,235,236,239] Lignin product yield and delignification degree are generally enhanced by raising the reaction temperature and the polarity of the reaction solvent,[231,234,236] although an optimum in solvent polarity can be found.[237] The presence of a Brønsted acid like H_3PO_4[238,244] or Lewis acids like $Yb(OTf)_3$ and $Al(OTf)_3$[240,241] also increases both delignification and monomer yield, enabling RCF at milder conditions.

Next to the reaction conditions, the lignin product yield depends strongly on the lignocellulose source. Various feedstocks have been tested in RCF, including hardwoods (birch, poplar, beech, *etc.*), softwoods (pine, spruce, *etc.*) and herbaceous crops (miscanthus, corn stover, *etc.*). To give a general overview of the monomer yields obtained with various feedstocks, the maximum reported monomer yield per lignocellulose substrate was collected from each RCF study (Table S1 in the ESI†), and with these data, histograms were constructed showing the monomer yield distribution for each biomass type, *viz.* hardwood, softwood and herbaceous crops (Fig. 14). These histograms display the number of data points within a certain yield interval.

As discussed in Section 2.1, the lignin composition and distribution of inter-unit linkages varies between feedstocks. To assess the influence of feedstock on reductive processing, Sels *et al.* performed RCF with various substrates (birch, poplar, miscanthus, and a spruce/pine mixture) and found a clear trend between the lignin S-content and the monomer yield.[231] Because the lignin S-content correlates with the β-ether content in lignin,[62,66] this observation indicates that a higher β-O-4 ether content in lignin results in a higher monomer yield. These findings were recently corroborated by Samec *et al.*, who showed a distinct trend between the monomer yield and content of native β-O-4 ether bonds in various woody substrates (birch, poplar, spruce, and pine).[352] In line with the results of Sels, Samec, and co-workers, the histograms in Fig. 14 show that the monomer yields generally decrease in the order of hardwoods > herbaceous crops > softwoods. For hardwoods, the majority of monomer yields are higher than 30 wt% and yields over 50 wt% are regularly reported, while the yields for softwoods are generally below 30 wt%. The yields for herbaceous crops mostly range from 20 to 40 wt%, although some outliers are present.[240,351]

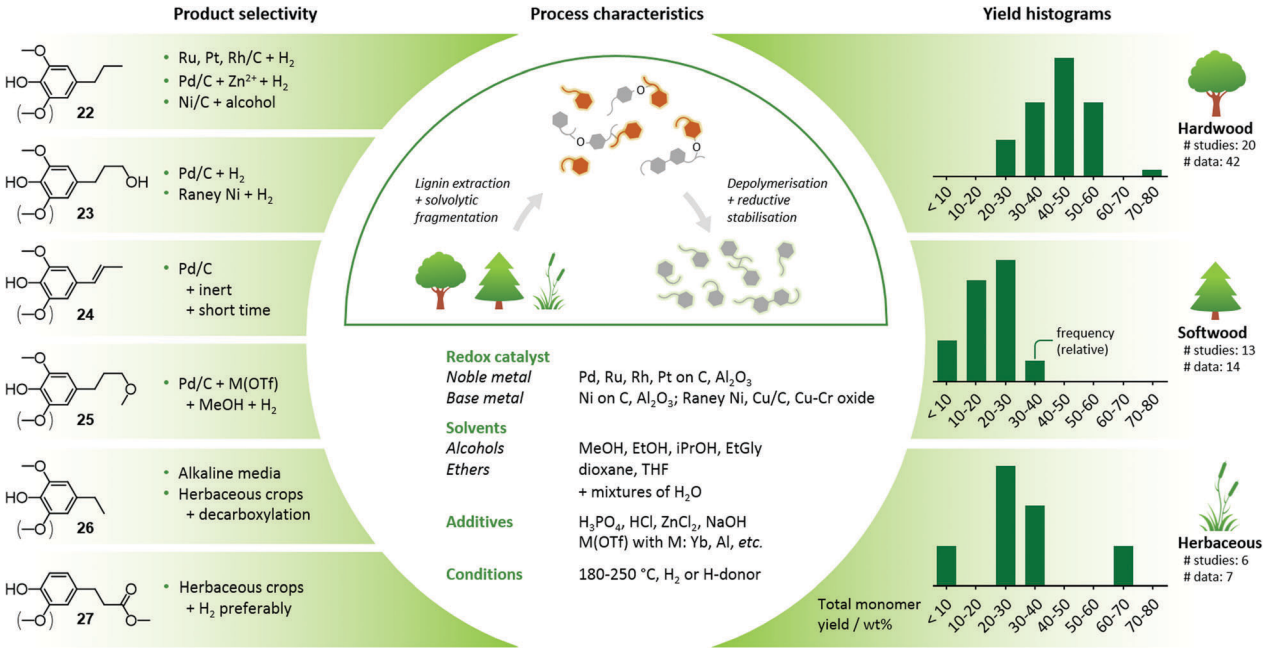

Fig. 14 Overview of reductive catalytic fractionation (RCF), highlighting the reaction conditions, attainable products, summary of the catalyst systems that enable the selective formation of each product, and histograms indicating the maximum reported monomer yields for each RCF study and for each biomass type.[64,195,226,231–245,342,351–366] The process characteristics and monomer yields of each individual study can be found in the ESI† (Table S1).

RCF is a highly selective process, with most studies indicating only a handful of phenolic products. The main phenolic monomers obtained from RCF are *para*-substituted methoxyphenols, with the structure of the substituent depending on the feedstock and process parameters. When starting from woody biomass, propyl-, propanol-, propenyl-, methoxypropyl-, and ethyl-substituted methoxyphenols can be selectively produced. Unique for herbaceous crops is that they can also generate propionic and acrylic acid-substituted methoxyphenols, which are either obtained as a carboxylic acid or ester (**27**) depending on the solvent used.[231,239,351] Similarly, *p*-hydroxybenzoate groups in feedstocks such as poplar and palm can be released as *p*-hydroxybenzoic acid, the corresponding ester, or the decarboxylation product phenol.[233,238,367]

Next to the feedstock, the substituent structure of the phenolic monomers mainly depends on the applied catalyst, additives, reaction time, and atmosphere (hydrogen or inert atmosphere). Fig. 14 provides an overview of the products that are obtained under certain process conditions. For example, propyl-substituted methoxyphenols (**22**) can be selectively produced under hydrogen atmosphere with Ru/C,[231,243] Pt/C,[244,353] Rh/C,[244] or a combination of Pd/C and a Zn-salt,[233,356] but also under inert atmosphere with a Ni/C catalyst.[235] In the latter case, an alcohol solvent is required as hydrogen-donor. These catalytic systems enable the effective removal of the γ-OH group, presumably through hydrogenolysis or tandem dehydration/hydrogenation. On the other hand, preservation of the γ-OH group is enabled by Pd/C[195,236–238,240,243,244,353,354,356,357] or RANEY® Ni[354,357] under hydrogen atmosphere, hereby selectively yielding propanol-substituted methoxyphenols (**23**). Interestingly, reaction with Pd/C under inert atmosphere generates propenyl-substituted methoxyphenols (**24**) at short reaction time, while the product selectivity shifts towards propyl-substituted compounds after extended reaction.[352] Hence, these observations suggest that pressurised H_2 is essential to effectively preserve the γ-OH group, whereas the reaction time should be considered as well to tune product selectivity. In addition, it has been shown that combining Pd/C with $Al(OTf)_3$ at high Al/Pd ratio selectively yields methoxypropyl-substituted methoxyphenols (**25**, upon reaction in methanol).[241] High amounts of metal triflate cause acid-catalysed etherification of the γ-OH group. A recent follow-up study by Hensen *et al.* showed that this etherification also takes place in presence of strong mineral acids such as HCl and H_2SO_4.[242]

Besides C_3-substituted compounds, ethyl-substituted methoxyphenols (**26**) can also be obtained, for instance by performing RCF under alkaline conditions (NaOH).[354,355] These compounds likely originate from β-O-4 hydrogenolysis and hydrogenation of the alkali-stable enol ether structures that are generated under alkaline conditions (Fig. 4).[238] As mentioned in the previous paragraph, RCF with herbaceous crops yields methoxyphenols with C_3-acid and -ester side-chains (**27**) in addition to other substituted methoxyphenols (**23–25**). Decarboxylation (and hydrogenation) of these side-chains also forms ethyl-substituted compounds (**26**).[239] Next to methoxyphenols, some RCF processes also produce ring-saturated compounds such as 4-alkylcyclohexanols, although usually in small amounts.[234,245,360,365] In most RCF processes however, the aromaticity is effectively preserved.

Closely related to RCF are reductive depolymerisation methods that perform conversion of the entire lignocellulose substrate in one-step. Unlike RCF, these methods do not perform a

fractionation, since both lignin and carbohydrates are converted and solubilised. Converting the entire lignocellulose in one step reduces the complexity of the biorefinery deconstruction process; however, it also shifts the separations burden downstream and potentially diminishes the versatility of the biorefinery as the fate of the carbohydrate and lignin fractions is intertwined. Only a few examples have yet been reported so far. Herein, the obtained products strongly depend on the applied catalytic system and process conditions. Li *et al.* demonstrated a one-pot reductive process with a carbon supported Ni–W_2C catalyst in water, in which the lignin fraction is converted into propyl- and propanol-substituted methoxyphenols and the carbohydrate fraction into sugar alcohols.[353] Alternatively, Ma *et al.* successfully converted various lignocellulose feedstocks into alkanes (hexanes and pentanes from cellulose and hemicellulose, respectively) and alkylated methoxyphenols (from lignin) over $LiTaMoO_6$ and Ru/C in aqueous phosphoric acid.[368] Ford *et al.* reported the one-stage conversion of wood sawdust in supercritical methanol into saturated alcohols.[369] In their process, the lignin and carbohydrate fractions were transformed into substituted cyclohexanols and C_{2-6} aliphatic alcohols, respectively, over a Cu-doped porous metal oxide (PMO). Recently, Wang *et al.* demonstrated the one-pot conversion of various lignocellulose feedstocks into alkanes over a Pt/$NbOPO_4$ catalyst in cyclohexane.[370] In this process, cellulose, hemicellulose and lignin are converted into hexanes, pentanes and alkylcyclohexanes (methyl-, ethyl-, isopropyl-, and *n*-propylcyclohexane), respectively. Remarkably, a high biomass conversion was obtained, although processing occurred at mild temperatures (190 °C) in a nonpolar solvent (cyclohexane), which is known to exhibit very poor lignin solubility.[235]

5.1.2 Non-reductive depolymerisation of native lignin. Next to reductive methods, effective depolymerisation of native lignin has also been demonstrated through thermal, oxidative, solvolytic, acid-catalysed, and base-catalysed methods. For example, in lignocellulose pyrolysis, up to 20 wt% of the native lignin can be converted into phenolic monomers under certain conditions (Section 4.2.3).[324–328] Also aerobic oxidation of woody biomass can yield over 30 wt% of phenolic monomers on lignin basis,[112,371] while even higher yields can be achieved through nitrobenzene or CuO oxidation.[337,372] Solvolysis of woody feedstocks, for instance with supercritical methanol or methanol/water mixtures, achieves up to 45 wt% monomer yields at very short reaction times.[373,374] In alkaline pretreatment of corn stover, 27 wt% of the native lignin could be transformed into phenolic monomers (Section 4.1.1).[144]

As discussed in Section 3.2, acid-catalysed depolymerisation of lignin generates highly reactive intermediates such as C_2-aldehyde-substituted phenolics and Hibbert's ketones which are prone to repolymerisation. Trapping of these intermediates has been extensively examined by Barta and other researchers in the depolymerisation of isolated lignins, through either acetal formation,[97,204,205,375,376] decarbonylation[97,377] or hydrogenation[97] (see Section 5.2.3). The acetal stabilisation strategy was successfully employed by Watanabe *et al.* in the acid-catalysed conversion of woody feedstocks, by using methanol as acetal forming agent.[378] Herein, monomer yields up to 3 wt% on wood basis (~10–13 wt% on lignin basis)[379] were achieved. Bruijnincx implemented decarbonylation (with a Rh complex) to trap the reactive intermediates in the Lewis-acid catalysed depolymerisation of isolated lignins (see Section 5.2.3) and native woody lignins, reaching 6 to 10 wt% monomer yields in the latter case.[377]

5.2 Depolymerisation of isolated lignin

Unlike methods focusing on native lignin, depolymerisation of isolated lignins is a much more investigated research topic, and has resulted in a large number of depolymerisation studies. In the following overview, the various studies are divided in four categories: (i) reductive, (ii) oxidative, (iii) base- and acid-catalysed, and (iv) solvolytic and thermal depolymerisation. The literature on a particular category can be further divided in several subsets, categorised by a specific depolymerisation method. For each depolymerisation method, process characteristics such as catalysts, solvents and reaction conditions are indicated in overview figures (Fig. 15, 17, 18 and 20). The structures of typical reaction products are shown as well, together with a graphical summary of the reported monomer yields, similar to the histograms shown in Fig. 14. Furthermore, each method is presented in context of monomer yield and product selectivity within the monomer fraction. The process characteristics and monomer yields of each individual study can be found in the ESI† (Tables S2–S13).

5.2.1 Reductive depolymerisation. During reductive depolymerisation, lignin is disassembled or hydrocracked in the presence of a redox catalysts and a reducing agent, which is almost exclusively hydrogen (Section 3.3). Hydrogen can either be present as hydrogen gas or can be derived from a hydrogen donating species, which is generally the solvent but can also be the lignin itself. When hydrogen gas is used, the process is called hydroprocessing. On the other hand, when hydrogen is derived from the solvent or lignin, it is termed liquid-phase reforming. In reductive depolymerisation, lignin is often not only depolymerised, but also deoxygenated by assistance of hydrogen (*via* hydrodeoxygenation or HDO). The degree of deoxygenation depends on the catalyst and process characteristics. The different reductive depolymerisation methods are summarised in Fig. 15. Hydroprocessing is subdivided into different methods, *viz.* (i) mild, (ii) harsh and (iii) bifunctional hydroprocessing. The degree of deoxygenation increases in the same order. Next to hydrogen, other reducing agents such as hydrosilanes,[349] Na,[153] and Zn[380] have been employed to depolymerise lignin. Reductive depolymerisation with hydrosilanes is briefly discussed below.

Mild hydroprocessing. Mild hydroprocessing is performed at relatively low temperatures (mostly ≤300 °C) and generates various *p*-substituted methoxyphenols (Fig. 15).[97,135,143,195,226,231,310,363,381–402] The mild conditions enable the preservation of the methoxy groups, in contrast to harsh hydroprocessing (*vide infra*). Mild hydroprocessing is performed in liquid-phase (water, an organic solvent, or solvent mixture) over a noble or base metal catalyst. The main methoxyphenol substituents are alkyl-groups (propyl-, ethyl- or methyl-) and propanol. Most studies report a rather high selectivity towards a handful of products.

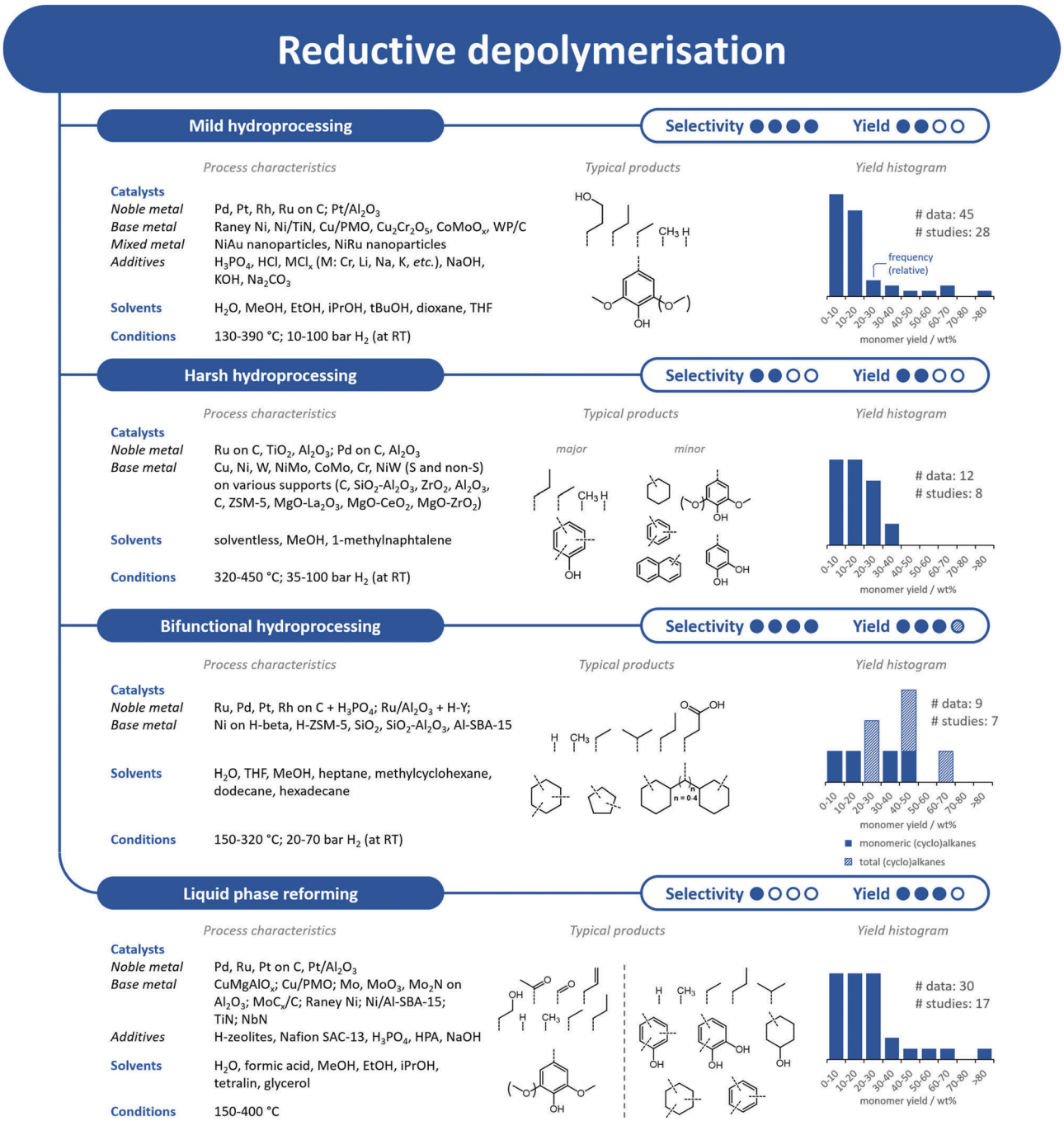

Fig. 15 Overview of the various reductive lignin depolymerisation methods with hydrogen as reducing agent: mild hydroprocessing,[97,135,143,195,226,231,310,363,381–402] harsh hydroprocessing,[403–410] bifunctional hydroprocessing,[178,281,310,411–415] and liquid phase reforming.[309,401,416–433] The yield histograms indicate the relative distribution of maximum reported monomer yields for each depolymerisation study and for each lignin substrate (if multiple substrates were used). The process characteristics and monomer yields of each individual study can be found in the ESI† (Tables S2–S5). Carboxylic acid groups in the products are esterified in case the process is performed in an alcohol solvent.

For example, Meier *et al.* showed that the monomer fraction from mild hydroprocessing is considerably less complex than that from fast pyrolysis, while both processes achieved similar monomer yields.[381] In general, monomer yields are below 20 wt%, although much higher yields have been occasionally reported.

The lignin structure has a pronounced influence on the monomer yield, but also on the monomer structure, as demonstrated by Bouxin *et al.*[143] They performed mild hydroprocessing of several isolated lignins (with Pt/Al_2O_3 in methanol/water at 300 °C) and found that conversion of a less condensed lignin, *i.e.* with a higher β-O-4 content, results in a higher monomer yield, but also in a higher retention of the C_3 side-chains (propyl, allyl, propanol, or methoxy-propyl) in the monomer fraction. The most reactive lignin, *viz.* poplar ARP lignin (Section 4.1.1),

yielded 14 wt% monomers. In a follow-up study, Bouxin, Bugg, and co-workers compared the reactivity of seven different lignins in two chemocatalytic and three biocatalytic depolymerisation processes, including mild hydroprocessing with Pt/Al_2O_3 at 300 °C. An overall correlation between the monomer yield and the β-O-4 content of the lignins was observed.[135] However, it was pointed out that this correlation is not fully conclusive and that other factors besides the β-O-4 content affect the monomer yield as well. In this particular study, oak dioxasolv lignin and miscanthus ionosolv lignin, both with a lower β-O-4 content than poplar ARP lignin (respectively 40 and 10% *vs.* 49% for poplar ARP lignin), were subjected to the same reaction, but generated much higher monomer yields (respectively 54 and 32 wt% *vs.* 14 wt%).

The destructive impact of a harsh lignin isolation procedure on the product yield was demonstrated by comparing the conversion of native birch lignin (see RCF, Section 5.1.1) and technical birch ethanosolv lignin over Ru/C in methanol at 250 °C.[231] While native birch lignin yielded 50 C% monomers, a yield of only 3 C% was obtained for the isolated birch lignin. In contrast, Torr *et al.* showed that a mild isolation procedure like enzymatic mild acidolysis hardly lowers the lignin reactivity, with the monomer yield being 22 wt% from native pine lignin and 21 wt% from pine EMAL in conversion over Pd/C in dioxane/water at 195 °C.[195] However, as mentioned in Section 4.2.2, EMAL is not an industrially relevant substrate. A recently developed lignin isolation method that is shown to preserve the lignin reactivity very well, is formaldehyde-assisted lignin extraction (Section 4.1.2, Fig. 10).[226] Luterbacher and co-workers demonstrated for various feedstocks that Ru/C-catalysed hydrogenolysis (in THF at 250 °C) of the formaldehyde-extracted lignins achieves very similar monomer yields on native lignin basis as direct Ru/C-catalysed hydrogenolysis of the native lignins (RCF in methanol at 250 °C). Starting from beech, spruce, and high-syringyl transgenic poplar (F5H-poplar), monomer yields on native lignin basis of 47, 21, and 78 mol% were obtained, respectively. For the F5H-poplar, the monomer yield was quantitative on extracted lignin basis. When beech lignin was isolated without formaldehyde, the monomer yield in subsequent hydrogenolysis was over 6 times lower, due to structural degradation.

Next to the structural properties of the isolated lignin, the product yield and selectivity during mild hydroprocessing is obviously also affected by the solvent, catalyst, and reaction conditions.[310,382–385,402] Furthermore, addition of co-catalysts such as Lewis acids ($CrCl_3$) or bases (NaOH or KOH) has been shown to enhance the monomer yields.[386–388] For example, Ma *et al.* found that the addition of a Lewis acid like $CrCl_3$ could increase the monomer yield from 7 to 29 wt% in the Pd/C-catalysed conversion of softwood alkali lignin in methanol at 260 °C.[386]

While mild hydroprocessing generally yields methoxyphenols, Barta *et al.* showed that this process can also selectively produce catechols.[389] By using a Cu/PMO catalyst in methanol, very high yields of propanol-, propyl-, and methoxypropyl-substituted catechol were obtained from candlenut organosolv lignin (up to 64 wt% based on isolated lignin at 140 °C), with a high selectivity towards propanolcatechol.

Harsh hydroprocessing. Harsh hydroprocessing involves lignin disassembly at high temperature (≥320 °C) and hydrogen pressure (≥35 bar), and mostly without a solvent, thus implicating reaction between a solid catalyst and the substrate.[403–410] This reaction is mainly studied with conventional CoMo and NiMo hydrotreating catalysts, but also noble metal and other base metal catalysts have been applied. At these high temperatures, the majority of methoxy groups are removed from the lignin products yielding phenol, methylated phenols (*e.g.* cresols, xylenols) and phenols with longer alkyl chains. Additionally, some mono- and polycyclic deoxygenated aromatics, alkanes, catechols, and methoxyphenols are also obtained (Fig. 15). Most studies indicate a rather broad product distribution with low selectivity towards individual products. Although most studies report yields below 20 wt%, higher yields are regularly obtained. For instance, Heeres *et al.* obtained over 22 wt% monomers from hardwood ethanosolv lignin (Alcell lignin) with either Ru/C, Ru/TiO_2, or Pd/C (at 400 °C without a solvent).[403] From pine kraft lignin (Indulin AT), Heeres, Barta, and co-workers achieved a 26 wt% monomer yield in solventless hydroprocessing with sulfided NiMo/$MgO–La_2O_3$ at 350 °C,[404] while a monomer yield of 35 wt% was obtained with sulfided NiW/C in methanol at 320 °C.[405]

Bifunctional hydroprocessing. In bifunctional hydroprocessing, lignin is converted into cycloalkanes by a bifunctional catalyst system containing both acid and metal sites (Fig. 15).[178,281,310,411–415] The acid sites catalyse hydrolysis (if water is present) and dehydration reactions, while the metal sites enable hydrogenolysis and hydrogenation. This process thus involves lignin depolymerisation and subsequent full HDO of the phenolic compounds, hereby funnelling a complex mixture of oxygenated compounds towards a small set of alkanes. Selective production of cycloalkanes from lignin has been demonstrated with Ni- and Ru-based catalysts, in water, alkanes or mixed water/organic solvents. The resulting cycloalkanes range from C_6 to C_{18} compounds, which are derived from both lignin monomers and dimers. Hence, it should be kept in mind that the total yield of cycloalkanes (in light blue in the histograms in Fig. 15) does not represent the actual monomer yield. Lercher, Zhao, and co-workers studied the conversion of beech organosolv lignin over Ni supported on SiO_2, H-ZSM-5, and H-beta in hexadecane, and found the cycloalkane yield (C_5–C_{14} cycloalkanes) to increase in that catalyst order.[411] While the latter two catalysts (Ni/H-ZSM-5 and Ni/H-beta) produced completely deoxygenated cycloalkanes, Ni/SiO_2 also yielded cyclic alcohols, which was attributed to the lower acidity of this catalyst. Furthermore, Ni/H-beta exhibited a higher selectivity to bicyclic alkanes than Ni/H-ZSM-5, which was explained by its larger pore size, enabling alkylation and condensation of phenolic monomers in the pores. At 250 °C, Ni/H-beta achieved a 35 wt% yield of cycloalkanes and a 42 wt% total hydrocarbon yield (gas and liquid), which could be increased up to 70 wt% at 320 °C. In another study by the same authors, an alkane yield of 46 wt% was obtained from corncob enzymatic hydrolysis

residue with Ni/SiO_2–Al_2O_3 in dodecane at 300 °C, with a very high selectivity toward monocyclic alkanes.[310] Interestingly, with Ni supported on less acidic oxides (like ZrO_2, MgO, Al_2O_3, or SiO_2) or in other solvents (like dioxane, benzene, or toluene), mainly phenolic products were obtained. Yang *et al.* also demonstrated the need for acid sites during lignin conversion to cycloalkanes over a combination of Ru/Al_2O_3 and H-Y.[178,412,413] With this catalyst system, a cycloalkane yield of 22 wt% was obtained from FT-DAP spruce lignin (Section 4.1.2).[178,414] In a follow-up study, it was found that the cycloalkane production could be enhanced by using the monometallic Ru/H-Y or bimetallic RuNi/H-Y as catalyst, with yields of 26 and 32 wt% respectively.[414] Rinaldi *et al.* used a bifunctional Ni/Al-SBA-15 catalyst in methylcyclohexane to convert poplar ethanosolv lignin, producing 45 wt% of volatile compounds at 300 °C, with over 99% selectivity towards cycloalkanes.[415]

Unlike the studies discussed in the previous paragraph, Lutherbacher *et al.* only reported monocyclic alkane yields (in dark blue in the histogram in Fig. 15).[281] They converted GVL-extracted corn stover lignin (Section 4.2.1) with Ru/C and H_3PO_4 in two steps, with the first step comprising THF/water at 150 °C and the second step heptane/water at 250 °C. Since corn stover-extracted lignin was used, also cyclohexanepropionic acid was obtained, which is derived from ferulic and *p*-coumaric acids. Ru/C outperformed other noble-metal catalysts like Pd/C, Pt/C, and Rh/C, yielding up to 38 C% monomers, which was only a little lower than the yield from native corn stover lignin (42 C%). The yield from GVL-extracted lignin could be increased to 48 C% by adding methanol in the second step, which stabilises carboxylic acids such as cyclohexanepropionic acid through esterification.

Liquid-phase reforming. Liquid-phase reforming of lignin is performed under inert atmosphere in a hydrogen-donating solvent or in presence of a hydrogen-donating agent (Fig. 15).[309,401,416–433] Well-known examples are tetralin, isopropanol, and formic acid, but also others such as methanol and ethanol possess the ability to donate hydrogen. Various noble metal (Pt, Pd, and Ru) and base metal (CuMgAlO_x, MoC, *etc.*) catalysts have been investigated for this reaction. While some studies report a small number of products, liquid-phase reforming mostly yields a large pool of compounds, such as methoxyphenols with a variety of alkyl- and oxygenated side-chains, catechols, alkylphenols, deoxygenated aromatics, and cycloalkanes. Monomer yields over 20 wt% are frequently reported and they can reach up to 86 wt%.

In the liquid-phase reforming of wheat straw soda lignin (Protobind 1000) over CuMgAlO_x, Hensen *et al.* found that ethanol as a solvent enables much higher monomer yields than methanol (17 *vs.* 6 wt% at 300 °C).[416,417] In contrast to methanol, ethanol hinders repolymerisation by (i) acting as a scavenger for lignin-derived formaldehyde and (ii) by capping phenolic units through etherification of the hydroxyl groups (O-alkylation) and alkylation of *ortho* positions (C-alkylation). By raising the reaction temperature to 380 °C, the monomer yield could be increased up to 60 wt%, with deoxygenated aromatics and cycloalkanes as predominant products.[417] Other commonly used lignin substrates, *viz.* Alcell lignin and softwood kraft lignin, could also be effectively converted, with monomer yields of 62 and 86 wt%, respectively. Li *et al.* performed liquid-phase reforming of softwood kraft lignin with MoC/C at 180 °C in various solvents (ethanol, methanol, isopropanol, and water), and also reached the highest monomer yield in ethanol (28 wt%; deoxygenated aromatics, methoxyphenols and benzyl alcohols).[418] Similar monomer yields could be obtained with Mo/Al_2O_3 (33 wt%) and Mo_2N/Al_2O_3 (28 wt%), while the yield was much lower with MoO_3/Al_2O_3 (5 wt%).[419,420] Heeres *et al.* were able to convert Alcell lignin into 48 wt% of monomers (deoxygenated aromatics, alkylphenols, catechols and alkanes) with Ru/C in a methanol/formic acid mixture at 400 °C.[422] In order to assess the effect of an acid co-catalyst in liquid-phase reforming, Lin *et al.* studied the conversion of bamboo enzymatic hydrolysis lignin in methanol/water at 270 °C over RANEY® Ni combined with acid zeolites.[421] Addition of zeolites like H-USY, H-ZSM-5 or H-beta raised the monomer yield from 13 up to 26–28 wt%. Weckhuysen *et al.* compared acid (H_2SO_4 and phosphotungstic acid) and alkaline (NaOH) additives in the Pt/Al_2O_3-catalysed liquid-phase reforming of pine kraft lignin (Indulin AT) in ethanol/water, and obtained the highest monomer yield with H_2SO_4 (18 wt%; mainly methoxyphenols).[401] From Alcell lignin, a monomer yield of 9 wt% was obtained. Furthermore, they also performed hydroprocessing of the kraft lignin with various catalysts (Pt/Al_2O_3, Pd/C, Ru/C, and Ni/SiO_2) in ethanol/water at 200 °C, and obtained considerably lower monomer yields compared to liquid-phase reforming (4–6 wt%).

Reductive depolymerisation with hydrosilanes. As an alternative to hydrogen, Cantat *et al.* demonstrated the reductive depolymerisation of lignin with hydrosilanes as reductant.[349,434] Lignin conversion was performed at room temperature under metal-free conditions, with an excess of Et_3SiH in CH_2Cl_2 in presence of $B(C_6F_5)_3$ as a Lewis acid catalyst. The process yields propyl- and propanol-substituted benzene-1,2-diol (catechol; from guaiacyl units) and benzene-1,2,3-triol (pyrogallol; from syringyl units), with all the hydroxyl groups being silylated (Fig. 16). Afterwards, the silyl-groups could be effectively removed through hydrolysis. By tuning the reaction time, and the hydrosilane and catalyst loading, the product selectivity could be steered towards either propyl- or propanol-substituted compounds. The authors performed depolymerisation of a range of formacell lignins (obtained through formic acid/acetic acid/H_2O pulping) from different woody feedstocks, and obtained monomer yields (corrected for the silyl groups) in the range of 11–41 wt% from hardwood lignins

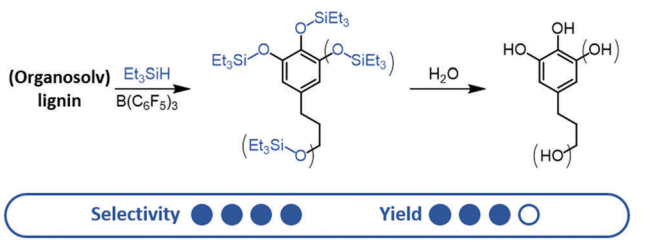

Fig. 16 Reductive lignin depolymerisation with hydrosilanes, followed by hydrolysis in a subsequent step.[349]

(oak, birch, beech, poplar, and hybrid plane) and 7–17 wt% from softwood lignins (pine, spruce, and cedar).[349] Next to formacell lignin, other organosolv lignins were prepared and tested in this depolymerisation reaction. Both the lignin isolation yield and monomer yield decreased in the order formacell > ethanosolv > methanosolv > acetone organosolv lignin, which shows that formacell pulping is an effective method to extract reactive lignin in high yield.

5.2.2 Oxidative depolymerisation. On the other side of the redox spectrum is oxidative lignin depolymerisation, which involves lignin conversion in presence of an oxidising agent. The main oxidants are dioxygen (referred to as oxygen here) and hydrogen peroxide, although others such as nitrobenzene and CuO are frequently applied for analytical purposes.[435] As discussed in Section 3.4, oxidative lignin depolymerisation can either induce cleavage of the side-chains, generating phenolic compounds, or cleavage of the aromatic rings, yielding aliphatic carboxylic acids. Most lignin oxidation studies focus on producing phenolic compounds (44 studies included in this review), whereas a much smaller amount targets aliphatic carboxylic and dicarboxylic acids (5 studies). Lignin oxidation towards phenolic compounds is mainly performed in alkaline media under oxygen or air, but it is also regularly studied in other media such as acidic or ionic liquids. An overview is presented in Fig. 17.

In the field of lignin oxidation, considerable research efforts have been devoted to the use of homogeneous metal complexes based on V, Cu, Co, Re, Mn, *etc.* as catalysts. Although their catalytic performance has been extensively examined on model compounds, studies on isolated lignins are scarce. Therefore, we would like to refer the interested reader to specialised reviews on

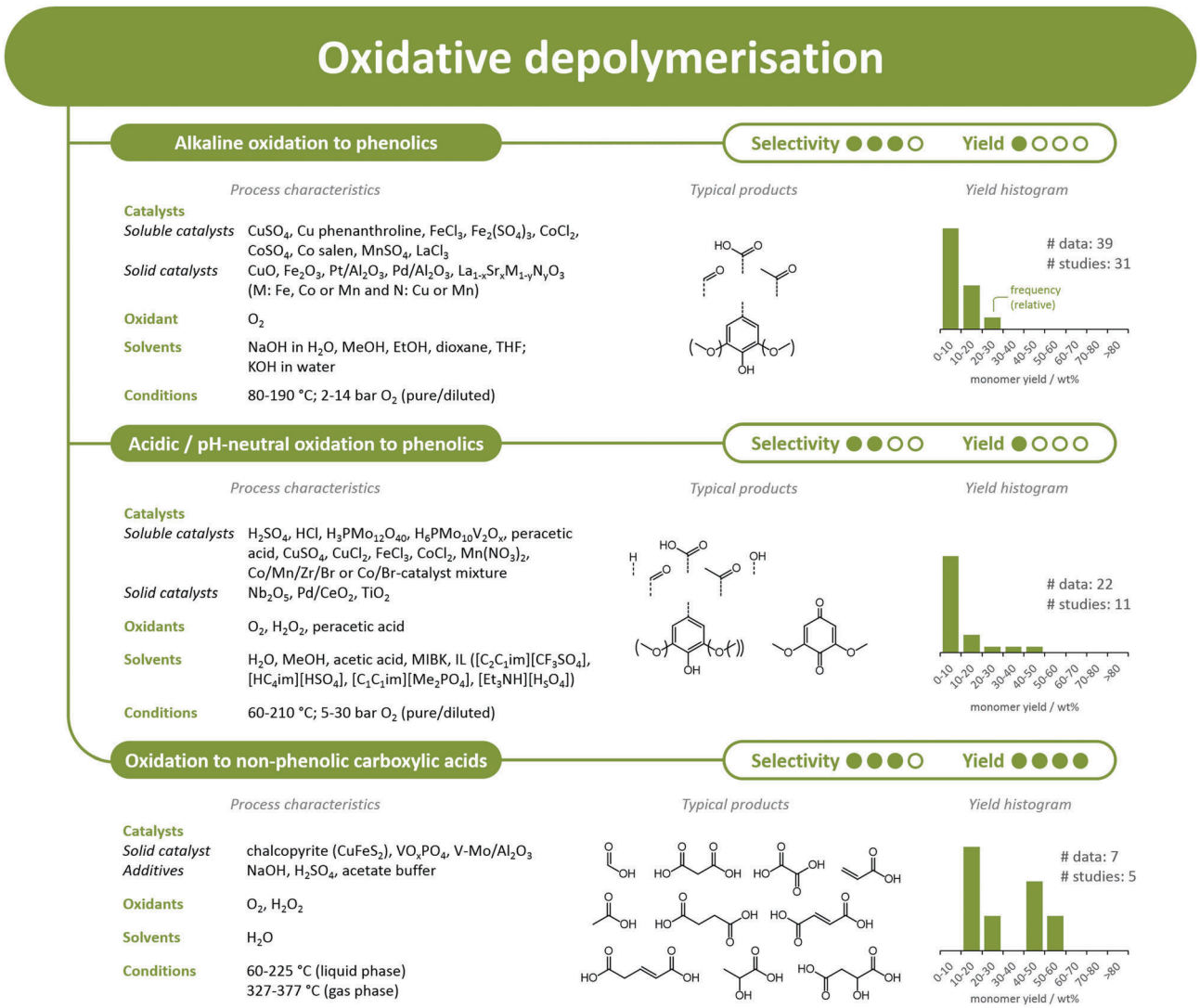

Fig. 17 Overview of the oxidative lignin depolymerisation methods: alkaline oxidation to phenolics,[112,113,180,338,443–474] acidic/pH-neutral oxidation to phenolics,[474–487] and oxidation to non-phenolic carboxylic acids.[179,488–491] The yield histograms indicate the relative distribution of maximum reported monomer yields for each depolymerisation study and for each lignin substrate (if multiple substrates were used). The process characteristics and monomer yields of each individual study can be found in the ESI† (Tables S6–S8). Carboxylic acid groups in the products are esterified in case the process is performed in an alcohol solvent.

this topic for more information.[23,24,108,436–440] Next to metal complexes and other homogeneous catalysts, also heterogeneous catalysts are frequently applied in lignin oxidation, both on model compounds and actual lignin, which is extensively discussed in dedicated reviews.[441,442]

Alkaline lignin oxidation to phenolic compounds. Since aerobic lignin oxidation in alkaline media enables the selective production of the aromatic flavouring agent vanillin, it has received considerable attention in academia and industry for almost a century. Commercial production of vanillin from lignin, with sulfite pulping liquor as lignin source, started in 1936, and even supplied 60% of the world market in 1981.[443] However, due to increasing environmental concerns, decreasing production of sulfite pulp (and liquor), and increasing manufacturing of synthetic vanillin from fossil resources, the production of vanillin from lignin has dropped, making up about 15% of the market today.[443,444] Lignin-derived vanillin is currently solely produced by the Norwegian company Borregaard, by oxidation of lignosulfonates.[443,444]

Alkaline lignin oxidation is almost exclusively performed with oxygen as oxidant (aerobic oxidation) in concentrated aqueous NaOH (0.5–4 M), with most studies using a 2 M NaOH solution.[112,113,180,338,443–474] Besided NaOH, also KOH[450] has been used as base. A high pH is necessary to (i) ionise the free phenolic hydroxyl groups in lignin, which facilitates oxidation (Section 3.4), and (ii) retard consecutive degradation of the aromatic aldehydes.[443,444] In addition, according to the mechanism proposed by Tarabanko *et al.*, a high pH is also required for deprotonation of certain reaction intermediates and nucleophilic addition of OH^- to the cinnamaldehyde-like intermediate before retro-aldol cleavage (Section 3.4 and Fig. S1B in the ESI†).[111–113,444] The atmosphere is either pure oxygen, diluted oxygen (with nitrogen), or air. The (partial) oxygen pressure and reaction temperature are usually in the range of 2–14 bar and 120–190 °C, respectively. The influence of temperature, oxygen pressure, and NaOH concentration on vanillin production from lignin has been extensively investigated by Rodrigues and co-workers.[444–448] They observed that increasing temperature or oxygen pressure accelerates both the formation and oxidative degradation of vanillin, and thus shortens the timespan to obtain the maximum vanillin yield. Furthermore, raising the reaction temperature was found to improve the maximum vanillin yield, which was not observed when increasing only the partial oxygen pressure.

Alkaline oxidation enables the selective formation of aromatic aldehydes (vanillin, syringaldehyde, and *p*-hydroxybenzaldehyde) from lignin (Fig. 7), with their distribution depending on the lignin source. Other products include aromatic acids (vanillic, syringic, and *p*-hydroxybenzoic acid) and acetophenone-derived compounds (acetovanillone and acetosyringone), and are depicted in Fig. 17. While fairly high monomer yields have been obtained from uncatalysed alkaline aerobic lignin oxidation, addition of a catalyst not only accelerates the reaction but in general also enhances the yield. However, some studies have observed no yield increase in catalysed runs.[445,451] A range of soluble and solid catalysts containing metals like Cu, Co, Fe, and Mn have been tested in this reaction, with $CuSO_4$ being most frequently used. A few examples of catalysts that have been shown to significantly enhance the monomer yield compared to non-catalysed runs are $CuSO_4$, either solely[180,449,452–455] or combined with $FeCl_3$,[180,454,455] Pd/Al_2O_3,[456–458] and Cu-doped perovskites based on Fe,[459] Co,[460,461] and Mn.[462–464] The monomer yields are mostly below 10 wt%, with yields in the range of 10–20 wt% being regularly reported. Only a few studies report yields over 20 wt%.[180,452,463]

Acidic and pH-neutral lignin oxidation to phenolic compounds. Acidic lignin oxidation has mainly been performed under aerobic conditions (with pure oxygen or air), either in diluted inorganic acids, or concentrated/pure acetic acid. Aerobic oxidation in diluted inorganic acids has been intensely studied by Von Rohr and co-workers.[474–478] They found that using methanol as co-solvent during lignin oxidation in acidic aqueous solutions considerably enhances the monomer yields, since methanol has the ability to quench reactive intermediates (it was suggested that methanol may react with intermediate carbenium ions and/or radicals) and reduce repolymerisation reactions.[474] Acidic oxidation of spruce[474,475] and pine kraft lignin[476,477] and softwood lignosulfonates[475] was shown to selective generate two products, *viz.* vanillin and methyl vanillate. The heteropolyacid (HPA) $H_3PMo_{12}O_{40}$ outperformed other acids such as H_2SO_4 and HCl in the production of these compounds, reaching combined yields of 9, 7 and 12 wt% from respectively spruce kraft lignin, pine kraft lignin (Indulin AT) and softwood lignosulfonates (Ultrazine NA) in methanol/water at 170 °C under pressurised oxygen.[474–476] In H_2SO_4-catalysed lignin oxidation, the yield of vanillin and methyl vanillate could be slightly enhanced by addition of metal salts like $CuSO_4$ and $CoCl_2$.[477] Interestingly, addition of $CuCl_2$ and $FeCl_3$ was found to strongly accelerate the formation of vanillin and methyl vanillate (without increasing the maximum yield), and also lead to the production of other monomers such as vanillin and vanillic acid with a methyl carboxylate group in the 5-position (methyl 5-formyl-2-hydroxy-3-methoxybenzoate and dimethyl 4-hydroxy-5-methoxy-1,3-benzenedicarboxylate). By performing continuous experiments, the authors also showed that high temperatures and short residence times are essential to reach high monomer yields, similar to alkaline lignin oxidation.[478]

Partenheimer studied aerobic lignin oxidation in concentrated and pure acetic acid with a Co/Mn/Zr/Br catalyst system.[479] This process selectively yields aromatic aldehydes (vanillin and syringaldehyde) and acids (vanillic and syringic acid), with the acids being the predominant products. Starting from a mixed hardwood acetosolv lignin, an 11 wt% combined yield of these compounds was reached at 180 °C in concentrated (92 wt%) acetic acid. Gonçalves and Schuchardt used a Co/Br catalyst system for aerobic oxidation of Organocell lignin (extracted with a methanol/water/NaOH/anthraquinone mixture) in pure acetic acid at 210 °C, which gave a 5 wt% combined yield of vanillin and vanillic acid.[480]

Ma *et al.* showed that peracetic acid as oxidant can effectively oxidise lignin in water at mild conditions (60 °C).[481]

Monomer yields of 18 and 22 wt% were obtained from alkali-extracted lignin from enzymatic residue of spruce and corn stover, respectively. These monomer yields could be further increased up to 35 and 47 wt%, respectively, by using a Nb_2O_5 catalyst. The main monomers were aromatic acids (vanillic, protocatechuic, *p*-hydroxybenzoic, and syringic acid) and *p*-hydroxylated phenol and guaiacol.

Lignin oxidation has also been performed in pH-neutral solvents such as methanol and ionic liquids. Aerobic oxidation of an organosolv lignin in methanol with Pd/CeO_2 yielded over 8 wt% monomers at 185 °C, with vanillin and *p*-hydroxybenzaldehyde as prevailing products.[482] Welton, Prado, and co-workers studied oxidative depolymerisation of ionosolv lignins from miscanthus,[483] willow,[484,485] and pine[484] in ionic liquids, and mainly obtained aromatic aldehydes, acids, and unsubstituted methoxyphenols as products. Although using oxygen as oxidant resulted in higher monomer yields than H_2O_2, the monomer yields were very low (<1 wt% from willow and pine ionosolv lignin in $[HC_4im][HSO_4]$ at 100 °C). Much higher monomer yields were reported by Wasserscheid *et al.* in the aerobic oxidation of beech organosolv lignin in $[C_2C_1im][CF_3SO_4]$ with $Mn(NO_3)_2$ at 100 °C (12 wt%)[486] and by Song *et al.* in the aerobic conversion of hardwood organosolv lignin in a $[C_1C_1im][Me_2PO_4]$/MIBK mixture with $CuSO_4$ at 175 °C (30 wt%).[487] While the latter process mainly yielded aromatic aldehydes, Wasserscheid *et al.* found that the product selectivity could be shifted from syringaldehyde as predominant product to 2,6-dimethoxy-1,4-benzoquinone by increasing the catalyst loading.

Lignin oxidation to non-phenolic carboxylic acids. Instead of producing phenolic compounds, lignin can also be oxidised into non-phenolic compounds such as small carboxylic and dicarboxylic acids (Fig. 17).[179,488–491] In this case, the phenolic compounds that are obtained as primary oxidation products are further converted into carboxylic acids (secondary products) through oxidative cleavage of the aromatic rings (Fig. 7 and Fig. S1, ESI†).[179,488,489] So, instead of aiming to minimise oxidative degradation of the phenolic compounds as in the previously discussed methods, these processes target their full conversion. This can be accomplished by applying reaction conditions under which the phenolic compounds are not stable (*e.g.* high O_2 pressure, long reaction times, reduced pH, *etc.*). Lignin conversion into carboxylic acids has mainly been conducted in liquid phase, namely in water under neutral,[489] acidic (H_2SO_4[179] or acetate buffer[488]) or alkaline (NaOH)[179,490] conditions with either H_2O_2 or O_2 as the oxidant at temperatures ranging from 60 to 225 °C. However, it has also been performed in gas phase.[491] In either case, the main products are formic, acetic, succinic, oxalic, and malonic acid, although also other acids have been obtained (Fig. 17). The reported monomer yields show a rather wide distribution, ranging from 11 to 56 wt%. Most processes show a high selectivity towards a handful of carboxylic acids.[179,488,489] Lee *et al.* have shown that lignin oxidation with H_2O_2 under alkaline conditions mainly yields oxalic and formic acid, while formic and acetic acid are the prevalent products under acidic conditions.[179] Starting from poplar FT-DAP lignin, they achieved a total carboxylic acid yield of 56 wt% under alkaline conditions (at 120 °C), compared to 41 wt% under acidic conditions (at 140 °C). Mae *et al.* performed lignin oxidation with H_2O_2 in pure water and obtained high yields of succinic acid, formic and acetic acid (total yield of 45 wt% from softwood kraft lignin oxidation at 200 °C).[489] In aerobic alkaline lignin oxidation, Demesa *et al.* observed oxalic and glutaconic acid as main products at lower temperature (up to 200 °C), while the product selectivity shifted to formic, acetic and succinic acid at higher temperature.[490] From softwood kraft lignin, a maximum carboxylic acid yield of 44 wt% was achieved at 225 °C. Ma *et al.* reached high selectivities for succinic and malonic acid in chalcopyrite-catalysed lignin oxidation with H_2O_2 in an acetate buffer solution.[488] At 60 °C, the total carboxylic acid yield amounted to 11 and 14 wt% from alkali-extracted lignin from enzymatic residue of spruce and corn stover, respectively.

5.2.3 Base- and acid-catalysed depolymerisation

Base-catalysed depolymerisation (BCD). Base-catalysed lignin depolymerisation is performed at high temperature (240–330 °C) in the presence of a soluble (mostly NaOH)[293,387,492–500] or solid base (Fig. 18).[501–504] The solvent is usually water, although (aqueous) organic solvents are also regularly used. The monomer yields are generally below 10 wt% and yields over 20 wt% have not been reported to the best of our knowledge. At relatively low temperature (≤300 °C), methoxyphenols are the prevailing products, which are either substituted (mainly aromatic aldehydes and acetophenone-derivatives) or unsubstituted.[293,492–494,501] At high temperature (≥300 °C), the selectivity shifts to catechol (generally the predominant compound) and alkylcatechols (methyl- and ethylcatechol).[293,493–496]

Lin *et al.* compared lignin BCD with fast pyrolysis, and found that the former generates a smaller set of compounds, with catechol and methylcatechol as predominant products.[496] Fast pyrolysis on the other hand yields a wide range of products, constituting substituted and unsubstituted methoxyphenols (Section 5.2.4). In the conversion of hardwood organosolv lignin with NaOH in water at 300 °C, Lercher *et al.* reached a 15 wt% monomer yield at very short reaction time (<5 min), after which it sharply dropped due to repolymerisation reactions.[492] Dhepe *et al.* tested a wide range of solid bases, comprising solid zeolites and oxides, in the conversion of softwood kraft lignin in ethanol/water at 250 °C, and obtained the highest monomer yield of 18 wt% with the basic zeolite Na–X.[502] Furthermore, the solvent has a large impact on the product yield and structure. For instance, Ma *et al.* showed that MgO-catalysed conversion of pine ethanosolv lignin in THF, methanol, ethanol, or an ethanol/water mixture resulted in much higher monomer yields than conversion in water (8–13 *vs.* 2 wt% at 250 °C).[503] In BCD of enzymatic lignin residues from corn stover with different hydrotalcite catalysts, Beckham *et al.* observed a high selectivity towards 4-vinylphenol when 3-methyl-3-pentanol was applied as a solvent, while the typical BCD products were obtained when operating in water.[501]

Acid-catalysed depolymerisation (ACD). Acid-catalysed lignin depolymerisation (ACD) is usually performed at high temperature

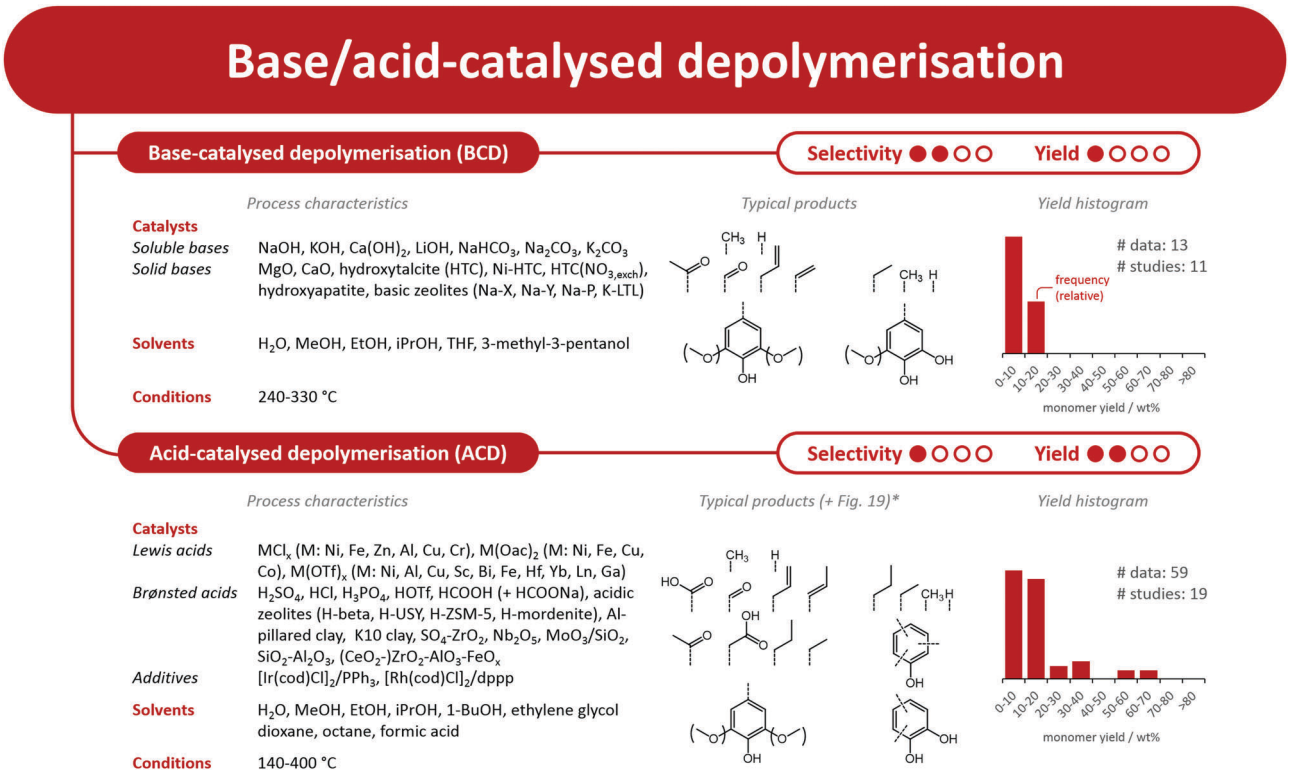

Fig. 18 Overview of base-catalysed[293,387,492–504] and acid-catalysed lignin depolymerisation.[97,204,205,375–377,386,432,505–516] The yield histograms indicate the relative distribution of maximum reported monomer yields for each depolymerisation study and for each lignin substrate (if multiple substrates were used). The process characteristics and monomer yields of each individual study can be found in the ESI† (Tables S9 and S10). Carboxylic acid groups in the products are esterified in case the process is performed in an alcohol solvent. *Specific products resulting from ACD in polyols are displayed in Fig. 19.

(≥250 °C), although lower temperatures around 140–180 °C are also often applied. Studies with model compounds have even shown that β-O-4 ether bonds can already be cleaved in ACD at temperatures as low as 85 °C.[93,94] A variety of soluble Lewis acids,[204,205,375,377,386,505–508] soluble Brønsted acids,[97,376,432,506,509–513] and solid Brønsted acids[509,514–516] have been utilised for this conversion, either in water, an organic solvent, or solvent mixture (Fig. 18). There's a broad distribution in reported monomer yields, ranging from very low (2 wt%) to very high (up to 61 wt%).

Thring, Ma, and co-workers studied Lewis acids like $NiCl_2$, $FeCl_3$, and $ZnCl_2$ for lignin conversion and found that these catalysts mainly generate non-substituted phenolic compounds.[505,506] A similar shift in product selectivity as in BCD was noticed from methoxyphenols to catechol with increasing temperature (from 250–260 °C to about 300 °C). Ma *et al.* tested various metal chlorides ($NiCl_2$, $AlCl_3$, $CuCl_2$, and $ZnCl_2$) in the depolymerisation of softwood kraft lignin in ethanol, and obtained the highest monomer yield with $ZnCl_2$.[506] At 300 °C, $ZnCl_2$ yielded 34 wt% monomers, with guaiacol and catechol as predominant products. Reaction in ethanol and other alcohols like methanol and butanol enabled much higher monomer yields compared to reaction in water, which was attributed to the higher solubility of the lignin products in alcohol solvents. Hensen *et al.* studied lignin conversion with metal chlorides and acetates at high temperature (400 °C), and observed low monomer yields and high char formation in water, while char formation was inhibited in ethanol, and higher monomer yields were achieved.[508] Conversion of wheat straw soda lignin (Protobind 1000) with $Cu(OAc)_2$, the best performing catalyst in their study, generated 13 wt% phenolic monomers in ethanol, compared to 6 wt% in water. The prevailing products in water were phenols and catechols, while reaction in ethanol mainly produced phenols and guaiacols. Also other products such as aliphatics, deoxygenated aromatics, ketones and acids were obtained, but these compounds are not included in the monomer yield since they are also partly or entirely derived from the ethanol solvent. Hence, a downside of the use of ethanol is its partial conversion under the applied reaction conditions. In a follow-up study, the use of an ethanol/water mixture was shown to significantly enhance the phenolic monomer yield in the $Al(OTf)_3$-catalysed conversion of the same lignin at 400 °C, increasing from 9 wt% in pure ethanol to 21 wt% in the solvent mixture.[507]

Dhepe *et al.* investigated lignin conversion with various soluble and solid Brønsted acids.[509,514] They observed a wide array of products, with the main compounds being methoxyphenols with oxygenated side-chains, like vanillin, guaiacylacetone and homovanillic acid. In pure water, no products were obtained, likely due to the low solubility of the lignin substrate (softwood kraft lignin).[514] In methanol/water however, depolymerisation with the mineral acids HCl and H_2SO_4 achieved monomer

yields of 29 and 39 wt%, respectively, from softwood kraft lignin at 250 °C.[509] A range of solid Brønsted acids were tested in the same reaction, with the highest monomer yields from H-ZSM-5, SiO_2–Al_2O_3, and H-USY (60, 59, and 55 wt% respectively). In contrast to Dhepe *et al.*, Ma *et al.* obtained monomer products from ACD of softwood kraft lignin in pure water. Depolymerisation with H_3PO_4 at 260 °C yielded 11 wt% monomers, with guaiacol being the predominant compound.[506]

While formic acid is frequently applied as a hydrogen-donor in liquid-phase reforming (in the presence of a redox catalyst, Section 5.2.1), it is also sometimes used without a redox catalyst and in this case primarily serves as a (weak) Brønsted acid catalyst.[432,510–513] Although a redox catalyst is generally required to release hydrogen from hydrogen-donors like methanol or ethanol, it is expected that formic acid also generates hydrogen in absence of a catalyst at high temperature. Therefore, formic acid-catalysed lignin conversion is likely not a purely acid-catalysed reaction, but also a reductive reaction, at least to a certain extent. Varying monomer yields have been reported for this reaction, ranging from 3 wt% in the conversion of wheat straw soda lignin (Protobind 1000) in ethanol at 360 °C (mixture of ethyl-, methyl- and unsubstituted phenol, guaiacol, and syringol)[512] to 33 wt% in the conversion of softwood kraft lignin in water at 246 °C (mainly catechol).[432]

Since effective acid-catalysed depolymerisation is generally hampered by repolymerisation of reactive intermediates, Barta, de Vries, Westwood and co-workers investigated various routes to quench these intermediates.[97] The C_2-aldehyde substituted intermediates formed during acid-catalysed depolymerisation (Fig. 5) were identified as the prime actors for undesired side reactions like condensation. It was demonstrated that these intermediates can be trapped by (i) acetal formation (with ethylene glycol, illustrated in Fig. 19), (ii) hydrogenation (with Ru/C), and (iii) decarbonylation (with an Ir complex). Especially the acetal formation route was found to be effective for stabilising monomers, as indicated by a threefold increase in monomer yield after addition of ethylene glycol in the triflic acid-catalysed conversion of walnut dioxasolv lignin. Furthermore, unlike other ACD methods, this particular process displayed a high selectivity towards a handful of products: C_2-acetals of phenol, guaiacol, and syringol.[97,376] Noticeably, although formaldehyde-assisted fractionation (Section 4.1.2; Fig. 10) is distinctly different from this depolymerisation method, it also relies on the reaction of a carbonyl group with a diol to form an acid-stable acetal, hereby preventing structural degradation. In a follow-up study on the ethylene glycol-assisted ACD process, the use of Lewis acid metal triflates as less corrosive and easier-to-handle alternatives to the strong Brønsted acid triflic acid was examined in the ethylene glycol-assisted depolymerisation process.[375] The best results were obtained with $Bi(OTf)_3$ and $Fe(OTf)_3$. These catalysts produced 15 and 19 wt% monomer yields, respectively, from walnut methanosolv lignin in dioxane at 140 °C, compared to 14 wt% with triflic acid. In a subsequent study, the organosolv pulping of beech, walnut and douglas fir with concentrated (95%) ethanol and butanol was examined, and the resulting ethanosolv and butanosolv lignins were subjected to ethylene glycol-assisted conversion with $Bi(OTf)_3$.[204] While butanosolv pulping enabled much higher isolated lignin yields than ethanosolv pulping (61–97 wt% *vs.* 16–20 wt%), the corresponding butanosolv lignins were less susceptible to depolymerisation. The butanosolv lignins from beech and walnut generated about 10 wt% monomers, compared to 17–18 wt% from the ethanosolv lignins. However, although these monomer yields are lower, the monomer yields on an initial lignin basis in the feedstock are higher due to the significantly higher isolated lignin yield of butanosolv pulping. Following this work, the conversion of a large set of self-prepared organosolv lignins and technical lignins (organosolv, soda, and kraft) was examined with $Fe(OTf)_3$ as catalyst.[205] A direct correlation was observed between the monomer yields and the β-O-4 content of the lignins. The self-prepared lignins enabled much higher monomer yields than the technical lignins (11–38 *vs.* 0.5–6 wt% respectively), due to their higher β-O-4 content. The highest yield of 38 wt% was obtained from mildly extracted methanosolv lignin from walnut. Again, butanosolv pulping enabled much higher isolated lignin yields than the other organsolv pulping methods, underlining its potential for lignin isolation.

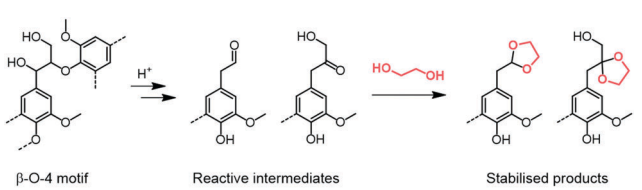

Fig. 19 Stabilisation mechanism during ethylene glycol-assisted ACD.[97,204,205,375,376]

Bruijnincx *et al.* investigated the Lewis-acid catalysed depolymerisation of lignin with metal triflates combined with decarbonylation of the C_2-aldehyde intermediates with a Rh complex.[377] Remarkably, next to stabilisation through decarbonylation, yielding methyl-substituted phenols, a second stabilisation pathway was observed, leading to the formation of propenylphenols. Although the mechanism of this additional pathway is unclear, the production of propenylphenols requires hydrogenation/hydrogenolysis to take place. The selectivity towards either methyl- or propenyl-substituted phenols could be controlled by varying the amount and strength of the Lewis acid catalyst. A higher acidity stimulated the decarbonylation route and thus enhanced the selectivity towards methyl-substituted compounds, but on the other hand slightly decreased the total monomer yield. Conversion of poplar dioxasolv lignin with the various Lewis acids (Yb, Sc, Ln, and $Ga(OTf)_3$) yielded 8 to 12 wt% monomers (in dioxane/water at 175 °C), compared to 10 wt% with the Brønsted acid triflic acid. Upon utilising triflic acid, propenylphenols were the main products.

5.2.4 Solvolytic & thermal depolymerisation

Solvolytic depolymerisation. Solvolytic lignin depolymerisation involves lignin conversion by solvolytic and thermal action. The solvent can be water,[432,495,501,506,508,517–523] an organic solvent,[309,383,386,387,416,499,501,503,506,508,519,524–527] or a solvent mixture.[509,513,517–519] Also hydrogen-donating solvents are regularly used (*e.g.* tetralin, isopropanol).[528,529] Processes using

formic acid are not included here, but are discussed under acid-catalysed depolymerisation (Section 5.2.3). The reaction temperatures range from 250 up to 450 °C. Lignin solvolysis generates a wide range of monomer products, with the product structure being influenced by both the solvent and reaction temperature. The overall product selectivity is usually low, although some more selective solvolytic processes exist.[432,520,521,524,525,528] The main monomer products are methoxyphenols, which are either unsubstituted or substituted with unsaturated (vinyl, allyl), saturated (methyl, ethyl), or oxygenated (aldehyde, ethanone, carboxylic acid) side-chains. Products with unsaturated and oxygenated side-chains are mainly obtained at lower temperature ($\leq$300 °C), while unsubstituted and alkyl-substituted compounds predominate at higher temperature ($\geq$300 °C).[517–519] Solvolysis in water or in a water/organic solvent mixture yields more unsubstituted and alkyl-substituted compounds than solvolysis in a pure organic solvents.[517,518] Furthermore, during lignin conversion in water at high temperature, methoxyphenols like guaiacol are converted to catechol, phenol, and cresols.[520–522,530] Solvolysis in hydrogen-donating solvents like isopropanol or tetralin mainly yields methoxyphenols with alkyl side-chains.[528]

The results on lignin solvolysis shown in Fig. 20 and Table S11 in the ESI† are either derived from studies that specifically focus on this process, or from studies on catalytic lignin depolymerisation (as the non-catalysed or blank runs). In most cases, solvolytic depolymerisation achieves lower monomer yields than catalytic (acid, base, or reductive) fragmentation, highlighting the benefits from implementing catalytic technology. The monomer yields in lignin solvolysis are generally below 10 wt%, although higher yields are sometimes reported. For instance, remarkable results were obtained in the conversion of softwood kraft lignin in water. At 350 °C, Sasaki *et al.* reached a monomer yield of 37 wt% with high selectivity towards catechol.[521] At a lower temperature of 265 °C, Onwudili *et al.*

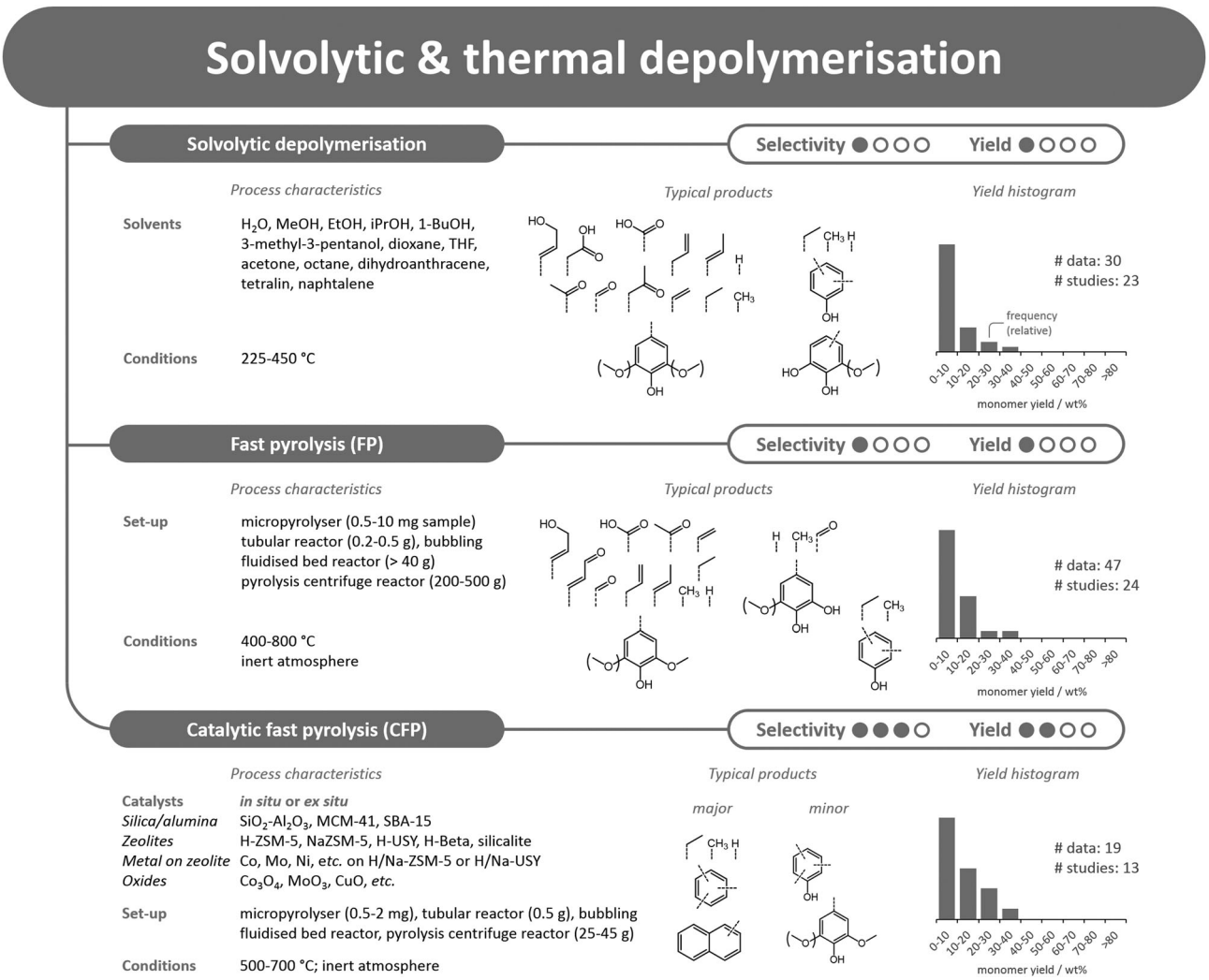

Fig. 20 Overview of solvolytic[309,383,386,387,416,432,495,499,501,503,506,508,509,513,517–529] and thermal lignin depolymerisation (fast pyrolysis[115,324,381,408,531–554] and catalytic fast pyrolysis[324,532,535–541,545,558–564]). The yield histograms indicate the relative distribution of maximum reported monomer yields for each depolymerisation study and for each lignin substrate (if multiple substrates were used). The process characteristics and monomer yields of each individual study can be found in the ESI† (Tables S11–S13). For solvolytic depolymerisation, the carboxylic acid groups in the products are esterified in case the process is performed in an alcohol solvent.

obtained 22 wt% monomers, with guaiacol as the main product.[432] Hu *et al.* extracted the lignin from hemicellulose-depleted corncob residue through an organosolv process with THF/water, and subsequently depolymerised the soluble lignin phase in THF at 300 °C.[524] A monomer yield of 24 wt% based on the lignin in the original corncob residue was achieved, with syringol, ethylphenol, and ethylguaiacol as the prevailing products.

Fast pyrolysis. Fast pyrolysis involves the (solventless) thermal decomposition of biomass by rapid heating to high temperature (450–600 °C) in the absence of oxygen.[319,320] The emerging pyrolysis vapours are immediately condensed into a bio-oil. Next to the bio-oil, char and gases are also produced. In lignin fast pyrolysis, the reaction temperature can range from 400 to 800 °C, but the highest liquid and monomer yields are generally obtained between 400 and 600 °C. Similar to lignin solvolysis, the results from lignin fast pyrolysis are derived from studies specifically on lignin fast pyrolysis, as well as from non-catalysed runs (blank runs) in catalytic fast pyrolysis (CFP) studies. Lignin fast pyrolysis reported in the academic literature is mostly performed at micro-scale (0.5–1 mg) in a micropyrolyser,[115,324,381,408,531–542] but also regularly at intermediate (0.2–0.5 g)[543–545] and multigram scale (>30 g).[408,531,546–554] Although most studies indicate monomer yields below 10 wt%, yields up to 20 wt% are frequently reported. Only very few studies report yields over 20 wt%. For instance, Van Bokhoven *et al.* and Zheng *et al.*, respectively, obtained monomer yields of 27 wt% (29 C%;[555] 650 °C)[535] and 30 wt% (at 700 °C)[532] from softwood kraft lignin. Guo *et al.* achieved monomer yields of 34–37 wt% (37–40 C%)[555] in the fast pyrolysis of rice husk pyrolytic lignins at 600 °C, whereas the yields from softwood kraft lignins under identical conditions were only 6–7 wt%.[545]

In fast pyrolysis, a bio-oil is obtained by rapid condensation of the pyrolysis vapours. Fast pyrolysis in a micropyrolyser is however mostly executed with direct GC analysis of the vapours and does not involve condensation. To investigate the influence of condensation on the pyrolysis products, Shanks *et al.* performed lignin fast pyrolysis with and without condensation of the vapours prior to GC analysis.[115] They observed that condensation results in significant repolymerisation of reactive phenolic compounds and thus lowers the monomer yield. This repolymerisation was furthermore found to be facilitated by acetic acid, a major product obtained from lignin fast pyrolysis. In line with these results, Bai *et al.* obtained a considerably higher monomer yield in the pyrolysis of corn stover acetosolv lignin in a micropyrolyser without condensation compared to pyrolysis at large scale in a fluidised bed reactor with downstream condensation (12 *vs.* 6 wt%).[549]

Fast pyrolysis, similar to solvolysis, produces a large pool of monomeric products, comprising substituted and unsubstituted methoxyphenols, catechols, and phenols, with the substituents mainly being unsaturated (mostly vinyl), saturated (methyl, ethyl), and to a lesser extend oxygenated (ethanone, aldehyde, carboxylic acid) groups (Fig. 20).[115,547–550] Meier *et al.* and Lin *et al.* demonstrated that fast pyrolysis generates a considerably more complex product mixture than mild hydroprocessing[381] and base-catalysed depolymerisation,[496] indicating the low product selectivity of fast pyrolysis. Shen *et al.* showed that compounds with unsaturated and oxygenated side-chains are produced at lower temperature (≤650 °C), while unsubstituted and alkyl-substituted compounds dominate at higher temperature (≥650 °C).[556] Also, a selectivity shift from methoxyphenols to catechols and phenols occurs by increasing the pyrolysis temperature. Evans *et al.* studied the pyrolysis of various native and isolated lignins, and found that the primary pyrolysis products from native lignins are coniferyl and sinapyl alcohol.[557] While pyrolysis of milled wood lignins rendered a similar product slate as native lignin, pyrolysis of organosolv and kraft lignin yielded smaller compounds such as unsubstituted and methyl-, ethyl-, and allyl-substituted methoxyphenols. This further exemplifies the chemically degraded state of technical lignins.

As briefly mentioned above, lignin fast pyrolysis has mainly been studied at very small scale in batch reactors, and realising large scale continuous pyrolysis has been shown to be challenging. In 2010, an international study on lignin fast pyrolysis was performed by fourteen laboratories, involving the fast pyrolysis of two lignin samples, one with high and the other with low lignin purity.[531] The study showed that (pure) lignin cannot be effectively pyrolysed in reactor systems designed for whole biomass, such as bubbling fluidised bed reactors. This is due to the low melting temperature of lignin and its tendency to agglomerate, causing plugging of the feeder and defluidisation of the reactor bed. Pyrolysis of the less pure lignin sample, with a high carbohydrate content, proceeded somewhat better than pyrolysis of the more pure lignin, since it behaved more like a typical raw biomass. It was concluded that new reactor designs will be required for continuous lignin fast pyrolysis. Bai *et al.* demonstrated that pretreatment of lignin with $Ca(OH)_2$ neutralises melting and agglomeration during fast pyrolysis and enables continuous pyrolysis of lignin in a fluidised bed reactor.[549] $Ca(OH)_2$ reacts with phenolic hydroxyl, carboxylic acid, and aldehyde groups in lignin, which are suggested to be responsible for the melting and agglomeration behaviour. Alternatively, De Wild *et al.* were able to effectively pyrolyse lignin in a bubbling fluidised bed reactor by pelletising lignin together with a natural mineral additive and by using a specially designed cooled-screw feeder.[546–548] Jensen *et al.* demonstrated that the problem of feeder plugging could be circumvented by using an alternative reactor set-up. The applied pyrolysis centrifuge reactor indeed prevented plugging of the feeder, but instead plugging of the cooling nozzle was observed after one hour of operation.[551]

Catalytic fast pyrolysis (CFP). In catalytic fast pyrolysis (CFP), pyrolysis is executed in presence of a catalyst, which is either physically mixed with the biomass in the pyrolysis reactor (*in situ* CFP) or is placed after the pyrolysis reactor and is only contacted with the pyrolysis vapours (*ex situ* CFP). Similar to lignin fast pyrolysis, published work in lignin CFP is mostly performed in a batch micropyrolyser with a very small amount of lignin (0.5–2 mg). Most studies using a micropyrolyser perform *in situ* lignin CFP with a high catalyst-to-lignin ratio (ranging from 2 to 20).[324,535–541,558–561] CFP in a micropyrolyser can also be run in *ex situ* mode, by separating the lignin and catalyst by

quartz wool in the pyrolysis tube,[532] or by placing the catalyst in a second microfurnace, which makes it possible to perform pyrolysis and catalytic conversion at different temperatures.[562] Lignin CFP at intermediate scale (0.5 g lignin) has been performed by Guo *et al.* in a tubular reactor (*ex situ* mode)[545] and at larger scale by Huber *et al.* in a fluidised bed reactor (*in situ* mode)[563] and by Jensen *et al.* in a pyrolysis centrifuge reactor (~30 g; *ex situ* mode).[564]

Most lignin CFP studies show a high selectivity towards deoxygenated aromatics such as benzene, toluene, xylene, and naphthalene. Next to these compounds, also some (methoxy)-phenols are usually produced (Fig. 20). Several studies have also indicated the formation of short chain olefins and alkanes.[558,562,564] The monomer yields (sum of deoxygenated aromatics and (methoxy)phenols) are generally below 20 wt%, with a few studies reporting higher yields.[535,541,545] Because deoxygenated aromatics are the major products, the monomer yields should be interpreted with caution, since these compounds can be produced from both lignin and carbohydrates. Several lignin CFP studies have been performed with impure lignins such as enzymatic or acid hydrolysis residue, in which the carbohydrates content can reach up to 50 wt%.[539,560]

Lignin CFP is mainly studied with acidic zeolites,[324,535–541,545,558–565] but also with other catalysts such as mesoporous silicas,[538,545] oxides,[535,536,538] and metal-supported catalysts (Fig. 20).[532,536,538,539] Van Bokhoven *et al.* indicated that the catalyst has a dual role in CFP, namely (i) the stabilisation of the intermediates, hereby preventing repolymerisation and cokes formation, and (ii) the transformation of depolymerised intermediates into the targeted products.[535] They showed that non-acidic porous catalysts like Na-ZSM-5 and silicalite only stabilise the intermediates, and thus yield the typical lignin fast pyrolysis products (phenols and methoxyphenols), while acidic catalysts like H-ZSM-5 further convert the primary products to deoxygenated aromatics. A clear increase in yield of deoxygenated aromatics was observed with decreasing Si/Al ratio, and thus increasing acid density, for a range of H-ZSM-5 catalysts. A similar trend was observed by Boateng *et al.* and Zhang *et al.* with both H-ZSM-5 and H-beta zeolites.[560,561] A mechanism for the conversion of methoxyphenols to deoxygenated aromatics over acidic zeolites was recently proposed by Brown *et al.*[566]

Lignin CFP is most often performed with H-ZSM-5, as this catalyst enables both a high monomer yield and deoxygenated aromatics selectivity.[541,545,560–562] Several studies have shown that H-ZSM-5 outperforms other zeolites such as H-Y and H-beta in terms of monomer production. For instance, Moutsoglou *et al.* compared H-ZSM-5 and H-Y in CFP of aspen butanosolv lignin at 600 °C, and obtained a higher monomer yield (28 *vs.* 17 wt%) and selectivity towards deoxygenated aromatics (82 *vs.* 35%) with H-ZSM-5.[541] Zhang *et al.* observed a higher monomer yield but lower selectivity towards deoxygenated aromatics with H-ZSM-5 compared to H-beta and H-Y.[561] This was ascribed to the small pores of H-ZSM-5, which hamper the conversion of bulky intermediates such as syringols to aromatics. In contrast, Van Bokhoven *et al.* reached a higher monomer yield (47 *vs.* 44 C%) and selectivity towards deoxygenated aromatics (83 *vs.* 77%) with H-USY compared to H-ZSM-5 in the conversion of softwood kraft lignin at 650 °C, which was attributed to the larger pore size of H-USY.[535] For more information on lignin fast pyrolysis and CFP, we refer the reader to more specialised reviews.[116,320,567–569]

5.2.5 Two-step lignin depolymerisation. In all previously discussed methods, lignin depolymerisation is performed in a single step. An alternative strategy is to perform a two-step process, wherein the goal of the first step is to weaken the β-O-4 linkage by altering the chemical structure of the motif. As a result, subsequent depolymerisation in a second step can occur at milder conditions, which favours the rate of depolymerisation over repolymerisation. Two primary methods to enhance the reactivity of lignin have been reported to date, namely (i) oxidation of benzylic alcohols and (ii) methylation of benzylic alcohols.

Benzylic alcohol oxidation and depolymerisation. Oxidation of the benzylic alcohol in β-O-4 lignin units significantly weakens the C_β–O ether bond[570] and thus facilitates subsequent depolymerisation.[571] Several groups have investigated a two-step depolymerisation strategy comprising benzylic alcohol oxidation on lignin β-O-4 model compounds.[380,572–582] Stahl and co-workers studied benzylic oxidation of a non-phenolic β-O-4 model compound with various stoichiometric oxidants and through metal- and non-metal-catalysed aerobic oxidation. The best result was obtained with a 4-acetoamide-TEMPO/HNO_3/HCl catalyst system under oxygen atmosphere.[572] This catalyst system was also able to selectively oxidise other phenolic and non-phenolic β-O-4 models, and even an actual lignin sample, namely aspen CEL (verified through 2D NMR). In a follow-up study, the authors demonstrated the effective depolymerisation of this oxidised lignin in an aqueous formic acid/sodium formate solution at 110 °C, yielding 52 wt% monomers, in contrast to only 7 wt% monomers from non-oxidised lignin.[573] The monomer fraction was composed of phenolic diketones (syringyl- and guaiacyl-1,2-propanedione, Fig. 21), aldehydes (syringaldehyde and vanillin), and acids (syringic, vanillic, and *p*-hydroxybenzoic acid). Alternatively, Westwood *et al.* achieved selective benzylic oxidation of non-phenolic β-O-4 models, a β-O-4 model polymer, and birch dioxasolv lignin with a DDQ/*t*BuONO catalyst system under oxygen.[380] Subsequent reductive depolymerisation of the oxidised lignin with an excess amount of Zn in presence of NH_4Cl at 80 °C yielded 6 wt% monomers, with a very high selectivity to syringyl-3-hydroxy-1-propanone. In following studies, this depolymerisation strategy was also applied to other lignins, generating 1 to 5 wt% combined yields of guaiacyl- and syringyl-3-hydroxy-1-propanone.[135,581]

Other examples of two-step benzylic alcohol oxidation and depolymerisation processes that have been shown to selectively convert β-O-4 model compounds are: (i) [4-AcNH-TEMPO]BF_4-mediated benzylic alcohol oxidation[574] or (ii) NHPI/2,6-lutidine-mediated electrocatalytic benzylic alcohol oxidation,[582] followed by photoredox-catalysed reductive β-ether bond cleavage with an iridium-complex, (iii) photocatalytic benzylic alcohol oxidation with Pd/$ZnIn_2S_4$ proceeded by photocatalytic β-ether hydrogenolysis with TiO_2,[575] (iv) $VOSO_4$/TEMPO-catalysed aerobic benzylic

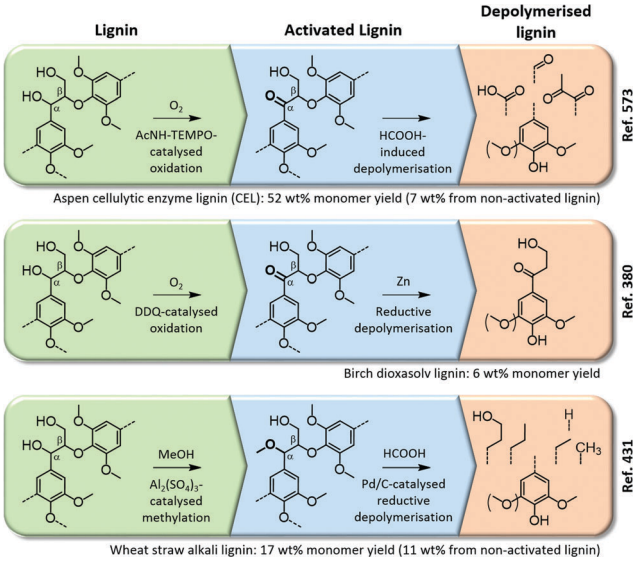

Fig. 21 Schematic overview of two-step lignin depolymerisation methods.

alcohol oxidation followed by aerobic oxidative C_α–C_β bond cleavage with a Cu/1,10-phenanthroline catalyst,[576] (v) Cp*Ir-catalysed oxidant-free benzylic alcohol dehydrogenation followed by reductive β-ether bond cleavage with Zn/NH_4Cl,[577] and (vi) benzylic oxidation with either TPPFeCl/*t*BuOOH, $DDQ/NaNO_2/O_2$ or $TEMPO/NaNO_2/NaCl/O_2$ followed by C_α–C_β bond cleavage through Baeyer–Villiger oxidation.[578]

Benzylic alcohol methylation and depolymerisation. Xu *et al.* and Ouyang *et al.* have both shown that methylation of the benzylic alcohol in a β-O-4 model considerably enhances its reactivity towards reductive depolymerisation.[364,431] Ouyang *et al.* showed that methylation of wheat alkali lignin (with methanol in presence of $Al_2(SO_4)_3$ under microwave irradiation) prior to Pd/C-catalysed liquid-phase reforming (in methanol/formic acid at 280 °C) enhanced the monomer yield from 11 to 17 wt%.[431] Xu *et al.* demonstrated that a Ni/C catalyst enables *in situ* benzylic alcohol methylation during lignin hydrogenolysis in methanol, and thus facilitates β-ether bond cleavage.[364] Organosolv pulping with alcohols is also known to etherify the benzylic alcohols within lignin,[64,204] which thus likely enhances the lignin reactivity.

5.3 Critical discussion on lignin depolymerisation

5.3.1 Monomer yield: impact of depolymerisation method. The histograms from Fig. 14, 15, 17, 18 and 20 are displayed as boxplots in Fig. 22 to allow for a visual analysis and comparison of the reported monomer yields for each depolymerisation method. At first glance, three depolymerisation strategies display a median (Q2) that exceeds an arbitrary threshold of 20 wt% monomers: reductive catalytic fractionation (RCF; Q2 of 37 wt%), bifunctional hydroprocessing towards cycloalkanes (32 wt%), and oxidation to non-phenolic acids (23 wt%). However, it should be noted that the cycloalkane yields reported for the majority of bifunctional hydroprocesses includes both mono- and bicycloalkanes. Unlike bifunctional hydroprocessing and oxidation to non-phenolic acids, RCF is the only method that generates aromatic products. Remarkably, more than 75% of the data points exceed the 20 wt% monomer threshold (see Q1). Amongst the other methods that result in aromatic products, especially liquid-phase reforming enables high monomer yields, with almost half of the data points over 20 wt% (Q2 of 19 wt%). Both Heeres *et al.*[422] and Weckhuysen *et al.*[401] compared lignin conversion through liquid-phase reforming and hydroprocessing, and reached considerably higher monomer yields with the former method. The highest monomer yields from liquid-phase reforming were reported by Hensen *et al.*, ranging from 60 to 86 wt% from various technical lignins (softwood kraft lignin, Protobind 1000, and Alcell lignin).[417]

Mild hydroprocessing (Q2 of 12 wt%), harsh hydroprocessing (12 wt%), ACD (12 wt%) and CFP (10 wt%) can be considered as the third most efficient depolymerisaton methods for production of aromatic monomers, with at least half of the data points over 10 wt%. The highest monomer yields through mild hydroprocessing have been achieved by Lutherbacher *et al.*, who even demonstrated the quantitative conversion of an isolated lignin.[226] The latter lignin was isolated through formaldehyde-assisted lignin extraction, which almost quantitatively retained the β-O-4 bonds in lignin. Harsh hydroprocessing can yield up to 35 wt% monomers, which was obtained by Barta *et al.* from softwood kraft lignin.[405] For ACD, yields ranging from 58 to 62 wt% were reported by Dephe *et al.* from industrial organosolv lignin and softwood kraft lignin.[509,514] Through CFP, Bokhoven *et al.* obtained about 34 wt% (47 C%)[555] monomers from softwood kraft lignin,[535] while Moutsoglou *et al.* and Guo *et al.* both reached monomer yields of about 28 wt% (39 C%)[555] from aspen butanosolv lignin[541] and rice husk pyrolytic lignin[545] respectively.

The last category includes the remaining depolymerisation methods, with the majority of data points below 10 wt%. In lignin oxidation, the highest yield was obtained by Lee *et al.* through alkaline aerobic oxidation (23 wt% from FT-DAP poplar lignin),[180] by Song *et al.* through aerobic oxidation in an ionic liquid (30 wt% from hardwood organosolv lignin),[487] and by Ma *et al.* through oxidation with peracetic acid (47 wt% from corn stover enzymatic residue).[481] For BCD, all reported monomer yields are below 20 wt%. Lignin solvolysis can yield up to 37 wt% monomers, which was obtained by Sasaki *et al.* through conversion of softwood kraft lignin in water.[521] Through fast pyrolysis, a maximum monomer yield of 37 wt% (40 C%)[555] was reported by Guo *et al.* from rice husk pyrolytic lignin.[545] As mentioned in Section 5.2.4, a large part of the data points from solvolysis and fast pyrolysis are derived from catalytic depolymerisation studies (on reductive depolymerisation, BCD, ACD and CFP) as the blank (non-catalysed) runs. In most cases, catalytic depolymerisation was found to outperform non-catalysed depolymerisation in terms of monomer yield, which illustrates the benefits gained from implementing catalytic technology.

In addition to these general observations, recently developed, sophisticated processes enable efficient lignin depolymerisation, such as (i) ACD with *in situ* stabilisation,[97,204,375–377] (ii) 2-step depolymerisation with benzylic oxidation,[380,573] and (iii) reductive depolymerisation with hydrosilanes.[349] For instance,

Barta *et al.* showed that by *in situ* stabilisation through acetal formation with ethylene glycol, the monomer yield from triflic acid-catalysed conversion of walnut dioxasolv lignin could be increased by a factor of three.[97] With $Fe(OTf)_3$ as acid catalyst, a maximum monomer yield of 38 wt% was achieved from walnut methanosolv lignin.[205] Stahl *et al.* demonstrated that benzylic oxidation of aspen CEL prior to depolymerisation with formic acid/sodium formate could enhance the monomer yield from 7 to 52 wt%.[573] In reductive depolymerisation with hydrosilanes, Feghali *et al.* reached monomer yields up to 17 wt% and 41 wt% from respectively softwood and hardwood formacell lignins,[349] which is in the same range of yields obtained through RCF.

As a critical note, it should be pointed out that the monomer yields from lignin depolymerisation strongly depend on three aspects, namely (i) the depolymerisation method, (ii) the lignin isolation method, and (iii) the lignin source (see Fig. 12 on lignin reactivity). This makes a clear quantitative comparison of the various depolymerisation methods difficult, as a broad range of different lignin substrates has been used. An additional hurdle is the large variation between reported results in literature, sometimes even between studies that use the same (or very similar) lignin and depolymerisation method. For instance, two studies on CFP of softwood kraft lignin in a micropyrolyser with H-ZSM-5 (silica-to-alumina ratios of respectively 25 and 30) at 650 °C indicated deoxygenated aromatics yields of respectively 5 wt%[559] and *ca.* 24 wt% (34 C%).[535] As another example, solvolytic conversion of softwood kraft lignin in water at 350 °C for 30 min has been stated to yield 4 wt%[519] and 37 wt%[521] monomers in two separate studies. Fortunately, a large set of data points (395) from an invaluable collection of depolymerisation studies (192) is available, which counterbalances part of the variation and makes it possible to evaluate the various depolymerisation methods. Nevertheless, for an even more accurate assessment of the different methods, studies are needed that examine multiple depolymerisation methods on the same lignin substrate and use similar analytical techniques to quantitatively verify the depolymerisation efficiency. Only in this way, the influence of the lignin structure can be decoupled from the depolymerisation method. For the same reason, studies that apply one specific depolymerisation method on a set of different lignins are as equally important to unambiguously evaluate the reactivity of different lignin substrates (*vide infra*).

In conclusion, the efficiency of depolymerisation is determined by the rate of lignin depolymerisation relative to the rate of repolymerisation or degradation. Two main principles can thus be followed to enhance the efficiency of lignin depolymerisation, namely (i) prevention of repolymerisation/degradation and (ii) enhancement of the depolymerisation rate. In Fig. 23, some specific strategies are outlined regarding these two principles.

Prevention of repolymerisation/degradation can for instance be accomplished by chemically converting reactive intermediates to more stable compounds. In reductive depolymerisation, this is performed by reductive stabilisation of reactive intermediates, which explains the high monomer yields that can be achieved (Sections 5.1.1 and 5.2.1). Stabilisation of reactive intermediates has also been demonstrated in ACD, either through acetal formation (with ethylene glycol[97,204,205,375,376] or methanol[378]), decarbonylation,[97,377] or hydrogenation.[97] In other depolymerisation methods without an active (chemical) stabilisation step, passive stabilisation is typically applied by physically removing the reactive intermediates from the reaction zone. This usually involves a precise tuning of the reaction time, by finding the right balance between depolymerisation and repolymerisaton/degradation. In methods like fast pyrolysis and CFP, the retention time of reaction products in the reaction zone is very short, and the products are rapidly cooled

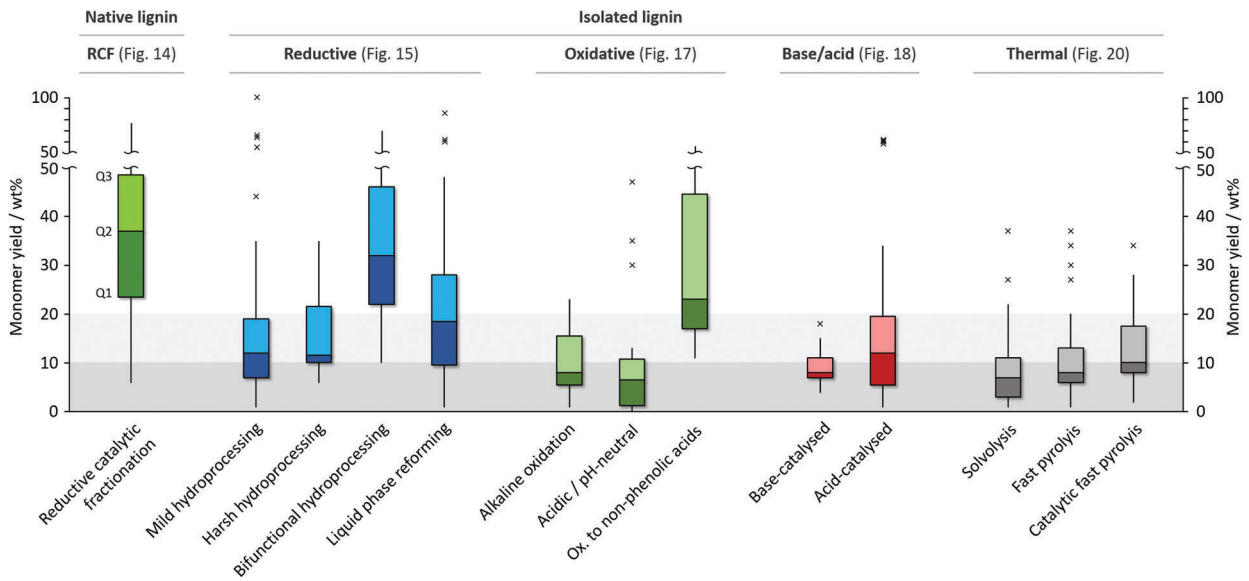

Fig. 22 Box plots showing the distribution of monomer yields for each lignin depolymerisation method. A total of 395 data points (gathered from 192 studies) are used. Data points exceeding the value of Q3 with more that 150% of the interquartile range (IQR = Q3–Q1) are displayed as outliers (×).

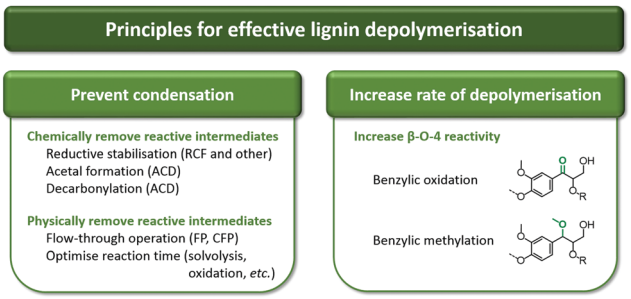

Fig. 23 Strategies to enable efficient lignin depolymerisation.

to minimise secondary reactions. However, it should be kept in mind that condensation of pyrolysis products also facilitates repolymerisation.[115] The need for short reaction times to maximise monomer yields was also clearly demonstrated in lignin solvolysis,[521] BCD,[492] and oxidation.[444,445] In ACD, Dephe *et al.* reported very high monomer yields, but it is however not clear how repolymerisation is prevented under their reaction conditions (250 °C in methanol/water for 30–120 min).[509,514]

As to the second principle, the rate of depolymerisation relative to the rate of repolymerisation/degradation can be enhanced by increasing the reactivity of cleavable linkages in lignin. This has been demonstrated by oxidation[380,573] and methylation[431] of the benzylic hydroxyl groups (*i.e.* 2-step approaches, Section 5.2.5).

While the principles outlined in Fig. 23 are based on the effective cleavage of ether bonds and prevention of carbon–carbon bond formation, an alternative strategy is to depolymerise lignin through scission of carbon–carbon bonds between the phenolic units. This includes both native carbon–carbon interunit linkages (Fig. 1) and carbon–carbon bonds formed during biomass fractionation (Sections 3.1 and 3.2). Research groups are increasingly focusing on the cleavage of carbon–carbon interunit linkages through oxidative pathways, and this has already led to various exciting results on model compounds.[583–585]

5.3.2 Monomer yield: impact of lignin structure. As indicated by several studies, the lignin β-O-4 content is one of the crucial factors that determines lignin's reactivity in depolymerisation reactions. For RCF, the impact of the β-O-4 content of native lignin on the monomer yield was directly demonstrated by Galkin *et al.*[352] and indirectly by Sels *et al.*,[231] and corroborated by the histograms displayed in Fig. 14 (see Section 5.1.1 for discussion). As a general trend, these histograms show that the monomer yields increase in the order softwoods < herbaceous crops < hardwoods, which correlates with their respective β-O-4 content. In order to relate the depolymerisation reactivity and the β-O-4 content of isolated lignins, Bugg and co-workers characterised seven isolated lignins and studied their chemo- and biocatalytic conversion.[135] They observed an overall positive trend between the catalytic results and the β-O-4 content of the lignins. Recently, Barta *et al.* examined the chemocatalytic conversion of a very large set of isolated lignins (27 lignins), and also observed an overall positive correlation between the monomer yields and the β-O-4 content of the lignins.[205] However, the fact that this relationship is not fully conclusive indicates that also other factors besides the β-O-4 content affect the susceptibility of lignin in depolymerisation reactions.

For instance, the presence of impurities can contribute to lignin's recalcitrance, and should not be overlooked.[21] Note that impurities can originate from (i) the biomass feedstock, (ii) chemicals used during biomass processing, or from external sources (*e.g.* metal leaching from reactor vessels and piping).[137] For example, sulfur originating from Na_2S in kraft pulping can act as a poison for many transition metal catalysts, thereby decreasing the catalyst life time.[137] Acid catalysts on the other hand are prone to deactivation by alkali and earth alkali metals (*e.g.* Na^+ and Ca^+) which exchange for H^+. As a third example, residual carbohydrates and degradation products (*e.g.* furfural) can decrease the activity of hydrogenation catalysts due to competitive adsorption.[242,586] For a more elaborate discussion on catalyst deactivation and strategies to resolve these issues, the interested reader is referred to a dedicated review by Lange.[137]

A statistical assessment of lignin depolymerisation suggests that, if a depolymerisation method only cleaves ether bonds, the maximum attainable monomer yield roughly equals the square of the fraction of cleavable inter-unit ether bonds, *i.e.* β-O-4 bonds.[21,244,349] Because the β-O-4 bond fraction in native lignin varies from about 45% to 75% for wood and herbaceous crops,[21,24,66,73] the monomer yields obtained through RCF, *viz.* 20–55 wt%, approximate the theoretical limit according to this mathematical relationship. The cleavage of β-O-4 bonds and formation of carbon–carbon bonds that occurs in many fractionation processes therefore has a detrimental effect on the efficiency of the subsequent depolymerisation step. Since technical lignins like softwood kraft lignin, Alcell lignin, and Protobind 1000 have low β-O-4 bond contents (below 10%),[75] the very high monomer yields that are obtained by certain liquid-phase reforming,[417,422,432] harsh processing,[405] ACD,[432,509,514] CFP,[535] and solvolysis processes[521] can only be explained by the ability of these methods to cleave carbon–carbon bonds.

While analytic methods such as 2D NMR and thioacidolysis make it possible to verify and compare the structural characteristics of lignins, valuable information regarding lignin reactivity can also be derived from depolymerisation studies. For instance, Bouxin *et al.* further evidenced the higher reactivity of ARP lignin compared to ethanosolv lignin *via* mild hydroprocessing.[143] In the same way, the higher reactivity of AFEX lignin compared to soda lignin was validated. Luterbacher *et al.* demonstrated *via* bifunctional hydroprocessing that GVL-extracted lignin is much more reactive than AAP lignin.[281] In other work, Luterbacher *et al.* showed that formaldehyde-assisted dioxasolv pulping retains the lignin reactivity much better than conventional dioxasolv pulping, as evidenced by results from mild hydroprocessing.[226] However, even with the same lignin substrates, different analytical and depolymerisation studies often lead to different conclusions. For instance, Hensen *et al.* obtained considerably higher monomer yields from commercial softwood kraft lignin than from Alcell lignin through liquid-phase reforming (respectively 86 *vs.* 62 wt%).[417] In line with this, Weckhuysen *et al.* also reached a higher monomer yield from commercial softwood kraft lignin compared to Alcell lignin in liquid-phase reforming (respectively 18 and 9 wt%).[401] Thring *et al.* and Barta *et al.* on the other hand found commercial softwood kraft lignin to be less reactive

than Alcell lignin, respectively in nitrobenzene oxidation (6 wt% monomers from softwood kraft lignin *vs.* 9 wt% from Alcell lignin)[587] and ethylene glycol-assisted ACD (0.5–2.6 wt% monomers from three softwood kraft lignins *vs.* 4.1 wt% from Alcell lignin).[205] De Wild *et al.* reached rather similar monomer yields through fast pyrolysis of both substrates (7 and 6 wt% from respectively softwood kraft and Alcell lignin).[546] These examples underpin the value of comprehensively evaluating lignin reactivity through multiple methods, as no universal approach exists.

Organosolv pulping is frequently considered to be a relatively mild isolation procedure that well preserves the reactivity of lignin, while traditional pulping methods such as kraft or soda pulping lead to highly condensed isolated lignins because of extensive degradation. A higher reactivity of organosolv compared to kraft lignin was for instance observed by Kaminsky *et al.* in the fast pyrolysis of beech and spruce ethanosolv and kraft lignins.[550] Nonetheless, as discussed in the previous paragraph, the reactivity of kraft lignin is often found to be higher than that of organosolv lignin. Furthermore, several studies have reported very similar results from kraft, soda, and organosolv lignins. For example, similar monomer yields were obtained from harsh hydroprocessing of softwood kraft and Organocell lignin (both 11 wt%),[409] from nitrobenzene oxidation of hardwood kraft and organosolv lignin (17 *vs.* 21 wt%),[468] and from liquid-phase reforming of wheat straw soda lignin (Protobind 1000) and ethanosolv lignin (19 *vs.* 16 wt%).[427] Hence, the assumption that organosolv lignins are always more reactive than kraft or soda lignins is an erroneous generalisation, as previously pointed out by Rinaldi *et al.*[21] Next to the applied fractionation method, the reactivity of the isolated lignin is strongly determined by the fractionation severity and biomass type, as indicated in Fig. 12.

Organosolv pulping can be performed with a wide range of aqueous organic solvents, with or without acidic additives and under varying reaction conditions (Section 4.1.2). Hence, large variations in process severity exist, which in turn affects the lignin reactivity. The variation in reactivity of different organosolv lignins is exemplified in several studies. Westwood *et al.* for instance showed that the alcohol solvent used in organosolv pulping strongly affects the reactivity, with the β-O-4 content and the monomer yield from ethylene glycol-assisted ACD being considerably higher for ethanosolv lignins compared to butanosolv lignins.[204] Furthermore, the β-O-4 content and monomer yield of their self-prepared beech ethanosolv lignin was much higher than that of a technical beech ethanosolv lignin (17 *vs.* 2 wt% monomer yield), which illustrates the large impact of different isolation conditions. Feghali *et al.* also demonstrated the effect of the pulping liquor on the reactivity of the organosolv lignin, with the monomer yield from hydrosilylation of pine lignins decreasing in the order formacell lignin (10 wt%), ethanosolv lignin (7 wt%), methanosolv lignin (5 wt%), and acetone organosolv lignin (2 wt%).[349]

Compared to lignin precipitates such as kraft and organosolv lignin, lignin-rich residues from enzymatic or acid hydrolysis are less frequently employed for lignin depolymerisation. Nevertheless, a few studies have examined their conversion. For instance, Barth *et al.* performed liquid-phase reforming on a range of softwood enzymatic and acid hydrolysis residues and obtained significantly lower monomer yields than from softwood kraft lignin (4–6 *vs.* 11 wt%).[424] On the other hand, nitrobenzene oxidation of corn stover enzymatic residues was found to yield 20–25 wt% monomers,[293] while Nb_2O_5-catalysed peracetic acid oxidation of spruce and corn stover enzymatic residue even generated up to 35 and 47 wt% monomers respectively.[481] The latter examples illustrate the potential of enzymatic hydrolysis residues as feedstock for aromatic monomers production. In analogy to isolated lignins, the reactivity of the lignin-rich residues is determined by the severity of the fractionation procedure and biomass type (Fig. 12). Within this context, the effect of residual carbohydrates or other impurities on depolymerisation efficiency should not be overlooked.

Lignin depolymerisation is frequently studied with commercial or technical lignins such as softwood kraft lignin (Indulin AT), Alcell lignin, and Protobind 1000, which are all derived from optimised fractionation processes and therefore represent industrially relevant lignin substrates. Next to technical lignins, also self-prepared lignins are regularly used in depolymerisation studies, in which their reactivity is assessed relative to other lignins. In most cases the isolated lignin yield (*i.e.* the yield of isolated lignin relative to the native lignin in the feedstock) is not indicated, despite the fact that the isolated yield is tightly linked with the lignin reactivity (Section 4.3.1). Reaching a higher isolated lignin yield requires a harsher or more extended isolation process, which in turn lowers the lignin reactivity. This was clearly illustrated by Pepper *et al.*, who showed that prolonged dioxasolv pulping of aspen wood increases the yield of isolated lignin, but decreases the reactivity of the isolated lignin in nitrobenzene oxidation.[588] Therefore, a fair evaluation of the effect of different isolation methods on the lignin reactivity requires similar isolated lignin yields. In analogy, the effect of enzymatic or acidic hydrolysis on the reactivity of a lignin-rich residue can only be fairly assessed if the lignin content in the residue is comparable, since removing more carbohydrates from the residue (and thus increasing the lignin content) requires more severe conditions or higher enzyme loadings, which in turn impacts the lignin reactivity.

Finally, we urge the lignin research community to consider the complete lignin mass balance when investigating (depolymerisation of) isolated lignins. Because lignocellulosic biorefineries aim at maximally valorising all lignocellulose constituents, the entire native lignin fraction should be taken into account in lignin depolymerisation research. When fractionation and depolymerisation are performed in separate stages, an optimal balance should be found between the lignin isolation and depolymerisation efficiency, *i.e.* between the isolated lignin yield and monomer yield, to maximise the product yield on a native lignin basis. Ideally, an isolation method should be used that enables high isolated lignin yields while maximally retaining their reactivity (see Section 4). As both the isolation and depolymerisation efficiency should be taken into account, a reduced lignin reactivity can be compensated by a higher isolation yield, and *vice versa*. This phenomenon was recently illustrated by Westwood *et al.* in

a comparative study between ethanosolv and butanosolv lignin from walnut shells and beech wood.[204] While the ethanosolv lignins were considerably more reactive than butanosolv lignins in ethylene-glycol assisted ACD, butanosolv pulping enabled much higher isolated lignins yields, resulting in over two times higher monomer yields on a native lignin basis.

5.3.3 Product selectivity. As illustrated in the above overview, the different depolymerisation methods also exhibit clear differences in terms of product selectivity. In the category of reductive depolymerisation, mild hydroprocessing is a selective method, as it yields a limited product slate consisting of substituted methoxyphenols. Especially mild hydroprocessing of native lignin (RCF) is highly selective towards a small number of compounds. Substituted methoxyphenols can be considered as primary depolymerisation products (Fig. 6), as the phenolic core of the products is retained. Harsh hydroprocessing on the other hand generates a more complex product mixture, since secondary reactions take place that change the phenolic core, *i.e.* demethoxylation, (de)alkylation, and hydrogenation of the aromatic ring. This yields a broad pool of primary (methoxyphenols) and secondary products ((alkyl)phenols, catechols, deoxygenated aromatics, *etc.*). In liquid-phase reforming, a similar shift from primary to a mixture of primary and secondary products can be observed with increasing process severity. Hence, the selectivity of harsh methods is generally low. Bifunctional hydroprocessing on the other hand enables a high product selectivity, as this method facilitates full defunctionalisation of the primary products, leading to one specific type of (stable) secondary products, namely cycloalkanes (concept of chemical funneling, Section 6). Summarising, selective (reductive) depolymerisation is achieved with methods that completely suppress secondary reactions (mild hydroprocessing, RCF) or with methods wherein secondary reactions occur to a maximal extent (bifunctional hydroprocessing). Methods situated in between yield a complex mixture of primary and secondary products (harsh hydroprocessing and liquid phase reforming).

While mild reductive depolymerisation usually generates substituted methoxyphenols, two unique processes in this category selectively produce substituted catechols and pyrogallols. Barta *et al.* showed that Cu/PMO under hydrogen atmosphere can selectively convert candlenut organosolv lignin to propanolcatechol and some other catechols,[389] while Feghali *et al.* presented a depolymerisation process using hydrosilanes as reducing agent which selectively produces either propyl- or propanol-substituted catechol and pyrogallol from woody organosolv lignins (Fig. 16).[349] In the former study, it is however not clear if the lignin structure (candlenut lignin) or the catalytic process is responsible for the formation of catechols.

In the group of oxidative depolymerisation methods, alkaline and acidic aerobic oxidation towards phenolic compounds are both selective towards a handful of products, with alkaline oxidation generating aromatic aldehydes, and acidic oxidation yielding a mixture of aromatic aldehydes and acids (or esters). Oxidation towards aliphatic carboxylic acids also usually generates a limited number of products.

Oppositely, in BCD, ACD, solvolysis, and fast pyrolysis, the overall product selectivity is relatively low. At relatively mild conditions, a wide variety of methoxyphenols is obtained, with various oxygenated, unsaturated, and saturated side-chains. Similar to reductive depolymerisation, moving from milder to harsher conditions shifts the product spectrum from methoxyphenols (primary products) to catechols and phenols (secondary products). In some cases, this improves the product selectivity. For instance for BCD, more severe conditions enable the rather selective formation of catechol and its methyl- and ethyl-substituted analogues.[293,493–496] Furthermore, moving from fast pyrolysis to catalytic fast pyrolysis enhances the product selectivity. The implementation of catalytic technology selectively steers the reaction towards secondary products, namely deoxygenated aromatics.

Two acid-catalysed depolymerisation methods that have recently been developed include an *in situ* product stabilisation step and generate a small set of products. The process reported by Barta, Westwood and co-workers selectively yields phenolic C_2-acetals,[97,204,205,375,376] while methyl- and propenyl-substituted methoxyphenols are obtained in the process disclosed by Bruijnincx *et al.*[377] The stabilisation mechanisms prevent the occurrence of unwanted repolymerisation reactions, thereby improving the monomer selectivity, as well as the total monomer yield.

The 2-step depolymerisation methods involving benzylic oxidation are selective towards a handful of compounds and also provide some unique monomers, which to the best of our knowledge have not been produced *via* traditional 1-step approaches. The process developed by Stahl *et al.* yields phenolic diketones next to aldehydes and acids,[573] while Westwood *et al.* obtained 3-hydroxy-1-propanone-substituted methoxyphenols with high selectivity (Fig. 21).[380]

Through specific depolymerisation methods, lignin can be selectively converted into a range of different compounds or categories (methoxyphenols, catechols, phenols, cycloalkanes, or deoxygenated aromatics). Interestingly, for substituted methoxyphenols, also the length of the side-chain can be tuned by selecting a suitable depolymerisation method. For instance, mild hydroprocessing selective yields C_3-substituted methoxyphenols, especially when applied to native lignin (RCF). Ethylene glycol-assisted ACD and alkaline RCF (mainly) generate C_2-substituted compounds, while C_1-substituted compounds are selectively formed through alkaline or acid oxidation, and ACD combined with decarbonylation.

Next to the depolymerisation methods, the product selectivity also depends on the lignin source and the fractionation method. As pointed out in Section 4.3.1, the reactivity of isolated lignin is determined by the fractionation method, with a more severe fractionation treatment generating a more condensed, less reactive lignin. Both Bouxin *et al.*[143] and Evans *et al.*[557] demonstrated that the lignin reactivity clearly affects the structure of depolymerisation products. They found, respectively through hydrogenolysis and pyrolysis, that more condensed lignins yield methoxyphenols with shortened side-chains, while the products from less condensed and native lignins better retain the original C_3 side-chains. This is also illustrated by the hydrogenolysis of native lignin (RCF), which almost exclusively generates products with C_3 side-chains.

Due to the impact of the lignin structure on the product distribution, the effect of the depolymerisation method on the product selectivity is most ideally examined using the same lignin substrate. This again points to the value of comparative studies.

Finally, valorisation of the obtained depolymerisation products strongly depends on the complexity of the product mixture. When a small number of compounds is produced, separation and purification of individual compounds may be feasible. Complex product mixtures on the other hand might be more suitable for applications that do not require pure compounds, but rather well-defined chemical and physical characteristics (precursors for resins, fuel-additives, solvents, *etc.*).[21,589–591] Alternatively, the complexity of depolymerised lignins can be diminished by means of convergent funneling approaches. The later strategy will be discussed in the next section.

6. Upgrading to targeted chemicals

A few of the monomeric compounds obtained from the various depolymerisation approaches (Section 5) can be applied as such in end-use applications, without further transformation. For instance, vanillin (from oxidative depolymerisation) can be used directly as vanilla fragrance and flavorant.[443,592] Nonetheless, many depolymerisation methods give rise to substituted phenolic compounds that require additional transformations *en route* to marketable chemicals. These down-stream transformations are grouped under the umbrella term upgrading, and include both chemocatalytic and biocatalytic approaches.

In addition, a frequently encountered hurdle that obstructs the implementation of lignin-derived compounds is the complexity of the lignin product mixture obtained from depolymerisation (Section 5.3.3). Isolating pure compounds from these often-complex streams is technically challenging, while the obtained quantities of a single compound are generally low. Although this methodology can be economically viable for certain high-value chemicals (*e.g.* vanillin),[443] a more energy and mass effective approach is needed for low cost drop-ins, as for instance fuels (*e.g.* specific alkanes, alcohols) and polymer building blocks (*e.g.* phenol, adipic acid, terephthalic acid). An alternative upgrading strategy to ease the problem of lignin product complexity and down-stream separation, is funneling. The main idea of this concept is to convert – *i.e.* funnel – a broad and heterogeneous mixture towards a smaller pool of central platform chemicals/intermediates, which in turn can be transformed into desired products. Inspired by Nature's inherent funnels from aromatic catabolism, the term was first introduced in the context of biological upgrading of lignin (Section 6.2).[145] Complementary, also certain chemocatalytic upgrading routes can be considered as funnels,[21] as will be explained in Section 6.1. Critical considerations on funneling are provided in Section 6.3.

6.1 Chemocatalytic upgrading of phenolic compounds

Lignin-derived monomers most often resemble their parent monolignol structure, and therefore consist of a phenolic core, substituted with one or two *o*-methoxy groups and a *p*-side-chain. The nature of the side chain strongly depends on the depolymerisation method, and ranges from simple alkyl-chains to highly functionalised substituents comprising alkenyl, alcohol, carbonyl, or carboxyl groups. In the following overview on chemocatalytic upgrading, a distinction is made between (i) transformations that affect the phenolic core and its substitution degree, and (ii) transformations that change the structure of the side-chain (Fig. 24). Regarding transformations that affect the phenolic core, the focus is on defunctionalisation through hydrodeoxygenation (HDO). HDO reactions result in a (partial) decrease of the product complexity and can therefore be considered as confluent, chemocatalytic funnels. After the HDO step, the resulting products can be subjected to secondary reactions that also affect the phenolic core. Therefore, the phenolic core transformations are subdivided in primary (HDO) and secondary (beyond HDO) transformations.

Next to HDO, also oxidation can be considered as a chemical route to funnel a mixture of phenolic compounds to a small set of compounds. For instance, Ma *et al.* showed that various phenolic compounds can be converted into a small number of carboxylic acids.[488]

6.1.1 Primary core transformations. Hydrodeoxygenation (HDO) of lignin depolymerisation products often forms an essential and primary upgrading step, because many applications (especially fuels) require compounds with reduced functionality and O/C ratio. HDO constitutes a highly active research field, and can be divided in four sub-domains based on the targeted products: alkanes, aromatics, phenols, and cyclohexanols (Fig. 24). They differ in (i) oxygen content (complete *vs.* partial HDO) and (ii) in the fate of the aromaticity (preservation *vs.* hydrogenation). Consequently, the four classes display a characteristic O/C and H/C ratio, as depicted in Fig. S2 of the ESI†. For a more elaborate discussion on HDO, the reader is referred to dedicated reviews on the topic.[23,24,66,71,321,323]

The class of lignin-derived alkanes, and more specifically C_{1-3}-alkylated cyclohexanes, includes interesting candidates to serve as mid-range fuel additives.[593] It has been demonstrated numerous times that cyclohexanes can be acquired in high yields from methoxylated phenols through the combined action of both metal catalysis (hydrogenation) and Brønsted acidity (dehydration and/or demethoxylation).[244,594–604] Noble metals (Ru, Rh, Pd, Pt) have been shown to provide excellent hydrogenation activity in these transformations, but also cheaper Ni-based catalyst have been applied successfully. The acidic counterpart of the bifunctional system can be provided by homogeneous acids (*e.g.* H_3PO_4, CH_3COOH, acidic IL) as well as heterogeneous acids (*e.g.* HZSM-5, HBEA). Note that this bifunctional approach can also be applied directly on isolated lignin (see bifunctional hydroprocessing; Section 5.2.1); hereby combining depolymerisation and upgrading in a one-pot process. In addition, alternative upgrading strategies have been proposed that rely on direct C–O hydrogenolysis instead of acid-mediated dehydration/demethoxylation.[605–607]

The second class of lignin-derived HDO-products comprises aromatic hydrocarbons (*e.g.* propylbenzene). In order to selectively acquire fully deoxygenated aromatics, direct C–O hydrogenolysis

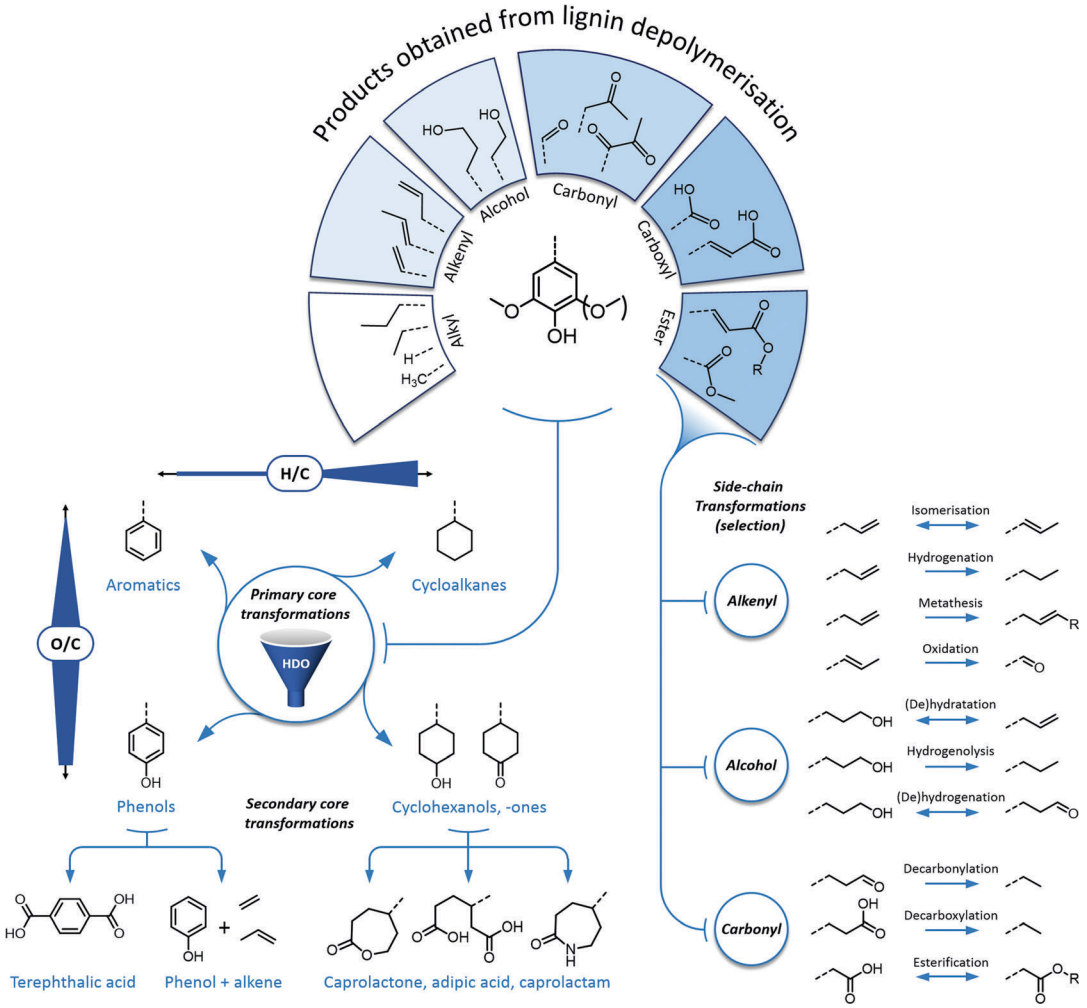

Fig. 24 Schematic representation of chemocatalytic upgrading strategies for substituted phenolics. Primary core transformations (*i.e.* hydrodeoxygenation) can decrease the mixture complexity and number of compounds, and are therefore considered as chemocatalytic funnels.

must be preferred over ring hydrogenation. This selectivity is favoured by operating in gas phase, at high temperature, and low hydrogen pressure (<1 bar).[608] Traditional sulfided CoMo and NiMo catalysts, which are widely used in the petrochemical industry for hydrotreating processes,[609] have been intensely investigated for this transformation.[66,71,323] However, these type of catalysts require co-feeding of sulfur (*e.g.* as H_2S) to compensate for sulfur losses and to maintain the catalytic activity.[608] Furthermore, demethylation reactions lead to catechols, which are sensitive to the formation of cokes under the employed conditions.[66,323] In search for alternatives, promising results have been obtained with non-sulfided catalysts such as MoO_3,[608] FeMoP,[610] Ru/TiO_2,[611] PdFe/C[612] and PtCo/C.[606] In addition to gas-phase configurations, several successful liquid-phase systems, with pressurised hydrogen (*e.g.* 20 bar) or H-donors, have been recently reported as well.[613–617]

Besides complete stripping of the monomer oxygen-functionality, upgrading can also aim to preserve the phenolic OH-group. For instance, selective demethoxylation of guaiacols gives rise to (substituted) phenols,[591] which can be used as intermediates for the synthesis of various new and drop-in polymer building blocks (*vide infra*). As such, higher value compounds are obtained from lignin, as opposed to the alkanes and aromatics. The reaction can be enabled by traditional sulfided CoMo- and NiMo-based catalysts.[323,618–620] Additionally, high yields of phenolics have also been obtained with supported noble metal (*e.g.* Pd/C, Ru/C),[611,612,621] and base metal catalysts (*e.g.* Fe/C, MoC_x/C, and WP/SiO_2).[612,622–624] It should be stressed that only a limited scope of phenolics has been studied thus far, mostly unsubstituted or alkylated guaiacols. However, an intriguing catalytic process was recently demonstrated by Zhu *et al.*, in which two complex functionalised methoxyphenols, namely vanillic acid and syringic acid, could be selective demethoxylated to *p*-hydroxybenzoic acid over a $MoWBO_x$/C catalyst.[625] A noteworthy illustration of how demethoxylation can be applied as a chemocatalytic funnel to convert a complex mixture of substituted methoxyphenols was recently presented by Rinaldi *et al.*[624] By employing a MoC_x/C catalyst, a pyrolysis bio-oil was converted and funneled primarily to (4-alkyl)phenols and aromatic hydrocarbons. Subsequently, these two major fractions could be easily separated from each other owing to their different polarity, which illustrates the benefits of the funneling concept for down-stream product purification.[624]

The fourth and smallest domain within the primary core transformations encompasses the partial HDO towards cyclohexanols *via* demethoxylation and aromatic ring hydrogenation, without removal of the alcohol functionality. This challenging task requires precise tuning of the catalyst properties and reaction conditions. Catalysts such as Ni/CeO_2,[626] $Ni/SiO_2–Al_2O_3$,[627] RANEY® Ni,[628] CoN_x/C,[629] $Ru/ZrO_2–La(OH)_3$,[630] $Ru–MnO_x/C$,[631] and Ru/C + MgO[632] have been reported as active and selective catalysts for this transformation in liquid phase.

6.1.2 Secondary core transformations. Upgrading can include extra transformations, downstream of the HDO funnel. These transformations are denoted as secondary core transformations, examples are depicted in Fig. 24. Alkylphenols,[591] obtained from demethoxylation of alkylguaiacol and syringol, can be selectively dealkylated with an acidic H-ZSM-5 zeolite, as disclosed by Verboekend *et al.*[633] Dealkylation of ethyl- and propylphenol leads to two commodity chemicals: phenol and short olefins (ethylene, propylene).[633] Another interesting drop-in chemical that can be obtained from lignin is terephthalic acid. Zhu *et al.* demonstrated that *p*-hydroxybenzoic acid, obtained from demethoxylation of vanillic and syringic acid, can be selectively carboxylated to terephthalic acid by $PdNiO_x/C$.[625]

Besides routes towards drop-in chemicals, complementary strategies to produce new polymer building blocks are also under investigation. For instance, alkylated cyclohexanols (*e.g.* propylcyclohexanol) can be oxidised towards the corresponding cyclohexanone.[626,634] Subsequent Baeyer–Villiger oxidation yields the alkylated caprolactone, a potential polyester building block.[626,634] Incorporation of a certain amount of alkylated caprolactone could alter the properties of regular polyester because of the presence of the alkyl side-chain. Analogously, alkylated cyclohexanone might be converted towards alkylated adipic acid or caprolactam.[626] These examples highlight that bio-based chemicals could provide an extra, distinguishing feature to the envisioned application, thereby unlocking new markets.

6.1.3 Side-chain transformations. In addition to transformations affecting the phenolic core, the side-chain can be targeted as well, for example through hydrogenation, hydrogenolysis (of –OH groups) and dehydration (Fig. 24). Such reactions can already take place during HDO, and can thereby contribute to a decrease of the product complexity. Alternatively, (other) side-chain transformations can also be performed separately, while leaving the phenolic core unchanged. The remaining of this paragraph summarises some selected examples.

Eugenol can for instance be isomerised to isoeugenol, which in turn yields vanillin upon oxidation. This route was already established in the late 19th century to satisfy the increasing demand for vanilla aroma.[443] Similarly, oxidation of *trans*-ferulic acid embodies a complementary route to vanillin.[635–637] Vanillin possesses the potential to serve as a versatile platform chemical, provided that cheap vanillin will become available from future biorefineries in large quantities, either *via* upgrading of (iso)eugenol or through oxidative depolymerisation of lignin. Numerous polymer building blocks can be derived from vanillin, as demonstrated by Caillol *et al.*[592,638,639] Possible vanillin transformations include the selective oxidation to vanillic acid, the partial reduction to vanillylalcohol,[640] and the reductive amination towards vanillylamine.[641,642] Analogous transformations and applications could be envisioned starting from syringaldehyde, but are far less studied.

Alternatively, unsaturated alkyl chains, such as for instance in (iso)eugenol and ferulic acid, can serve as anchoring point for olefin metathesis.[232] Bruneau *et al.* demonstrated the cross metathesis of eugenol with methylacrylate, acrylonitrile and acrylamide, leading to polyfunctional alkenes.[643] The reaction was mediated by Grubbs second generation type catalysts. Similarly, self-metathesis of eugenol provides a strategy to synthesise alternative bisphenols.[644,645] These and other lignin-derived bisphenols could become renewable substitutes for controversial bisphenol A.[646–649]

6.2 Biocatalytic upgrading of phenolic compounds

In addition to the aforementioned chemocatalytic funneling approaches, microbial transformations also offer promise for upgrading of lignin-derived products to value-added chemicals. The foundational concepts of biological funneling of lignin primarily arose *via* the elucidation of microbial aromatic-catabolic pathways, motivated in part by bioremediation studies of xenobiotic aromatic compounds in the biosphere.[650–653] Specifically, the genomes of many common microbes contain large batteries of enzymes that defunctionalise (*i.e.* funnel) aromatic compounds, including dimers and oligomers, into central intermediates, which are most commonly catechol, protocatechuate, and gallate in aerobes (Fig. 25).[654–658] These central intermediates can then be ring-opened *via* dioxygenase enzymes,[659–662] and eventually channelled into central carbon metabolism. We note that other aromatic-catabolic pathways exist, for example, for anaerobic assimilation of aromatic compounds, but these have not been extensively investigated to date for lignin conversion.[650] Overall, these natural aerobic pathways offer promise for the conversion of heterogeneous lignin streams to single compounds. Here, we briefly review several highlights of the emerging concept of biological funneling, and refer the reader to recent, detailed reviews for a more comprehensive treatment of this evolving area.[663–665]

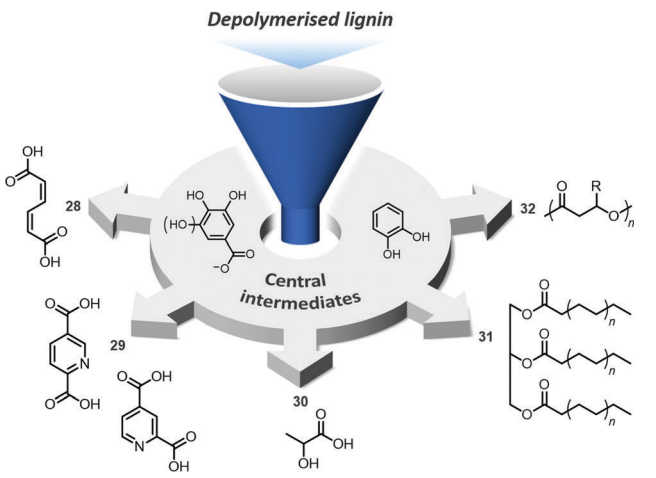

Fig. 25 Schematic representation of biocatalytic funneling strategies.

In the context of lignin upgrading, much of the initial work to use microbial catalysts was reported by Masai, Fukuda, Bugg, Eltis, and co-workers. In particular Masai, Fukuda, and colleagues have elucidated monomer and dimer catabolic pathways and enzymes in *Sphingobium* sp. SYK-6, which is able to cleave β-O-4 and 5-5 linkages in lignin dimers, as well as catabolise many common lignin-derived aromatic compounds.[658,666,667] Indeed, SYK-6 has become one of the most prominent model organisms for studying aromatic catabolism related to lignin.[668–670] Bugg, Eltis, and co-workers reported one of the first metabolically engineered microbial strains (*Rhodococcus jostii* RHA1) to convert lignin-derived aromatic compounds into a value-added chemical, namely vanillin, albeit at low titers from whole wheat straw, most likely due to the difficulty in depolymerising solid lignin with a bacterium alone.[671] Vanillin production was accomplished *via* the knockout of the vanillin dehydrogenase gene.

In the last several years, additional studies have been reported using aromatic-catabolic microbes to convert lignin-derived streams to value-added chemicals. In 2014, *Pseudomonas putida* KT2440 was employed to upgrade aromatic compounds present in alkaline pretreatment liquor (APL, Section 4.1.1) from corn stover to medium-chain-length polyhydroxyalkanoates (PHAs, **32**), which can be used directly as biodegradable plastics or thermally depolymerised into chemical building blocks.[145] This report coined the term biological funneling, and was among the first studies to use partially depolymerised lignin as a substrate – likely a requirement for effective microbial lignin upgrading – for producing a value-added product. Subsequently, *P. putida* was engineered to produce *cis*,*cis*-muconic acid (**28**), which is the *ortho*-cleavage product of catechol (catalysed by the 1,2-catechol dioxygenases), *via* deletion of the CatBC genes.[146] Muconic acid is a precursor to adipic acid, terephthalic acid, and other functional replacement products.[672] Continued developments for muconic acid have led to titers in excess of 30 g L^{-1} from lignin model compounds such as *p*-coumarate, ferulate, and benzoate at near-theoretical yields.[673–675] In a study from Bugg and colleagues, the authors demonstrated the production of novel pyridine dicarboxylic acids (**29**) from lignin-derived aromatic compounds in *R. jostii*. These pyridine dicarboxylic acids will likely exhibit novel chemical and polymer properties.[676] It was also shown by Johnson *et al.* that employing *meta* and *ortho*-cleavage pathways of protocatechuate and catechol, two key central intermediates, result in different pyruvate, succinate, and acetyl-CoA yields, thus affecting product yields of products derived from central carbon metabolism (*e.g.* lactic acid (**30**) from pyruvate).[677]

Multiple bacterial strains have also been compared directly in terms of their efficacy for biological funneling. Salvachua *et al.* conducted a comparative study of 15 bacteria on corn stover APL, comparing conversion and examining multiple properties of the lignin substrate during microbial conversion.[678] Four strains, *R. jostii* RHA1, *P. putida* KT2440 (and mt-2), *Amycolatopsis* sp., and *Acinetobacter baylyi* ADP1 all perform reasonably well, converting up to 30% of the lignin in a process-relevant stream to microbial biomass and, when deprived of excess of nitrogen, fatty acids, or PHAs. This study also demonstrated that these aromatic-catabolic microbes are able to secrete ligninolytic enzymes and partially depolymerise lignin oligomers, suggesting a conceptual link to the well-studied Consolidated Bioprocessing paradigm for microbial polysaccharide conversion.[678] In addition, Yuan and co-workers examined 15 strains of *P. putida*, comparing growth on vanillic acid.[679,680] Using a newly characterised *P. putida* strain (A514), the authors conducted a proteomics investigation to highlight the enzymatic pathways responsible for aromatic catabolism, engineered in the ability for enhanced secretion of an effective ligninolytic enzyme, and modified PHA biosynthesis to increase product titers.[677]

Yuan, Ragauskas, and colleagues have also conducted a significant body of work in an oleaginous bacterium, *R. opacus*, which is able to convert lignin-derived aromatic compounds into fatty acid-derived compounds (**31**) under nutrient deprivation.[680] For example, the authors have combined exogenous enzymes (*e.g.* laccases) with *R. opacus* and demonstrated a significant increase in lipid yields *via* extracellular lignin depolymerisation and uptake of released aromatic compounds.[680,681] Similar work was done with *P. putida* KT2440 in the presence of an entire fungal ligninolytic secretome, but only minor improvements in microbial biomass were observed.[682] Further understanding of the synergy between ligninolytic enzymes and aromatic-catabolic microbes is clearly needed to design microbial solutions that can attack oligomeric or polymeric lignin substrates effectively.

6.3 Funneling: critical considerations

Although upgrading of phenolic compounds has been intensely studied, the concept of funneling is still quite nascent. Multi-disciplinary research is needed to further improve its industrial relevance and to evaluate its feasibility. From the substrate side of the funnel, maybe the most important consideration in the subdomain of chemocatalytic upgrading is that the vast majority of research has been performed on pure, simple model compounds like phenol, cresol, anisole, and guaiacol. It will be essential to broaden the applicability of chemocatalytic upgrading routes in the near future by shifting to more complex compounds (substituted guaiacols and syringols), to compound mixtures mimicking depolymerisation streams, and of course, to raw depolymerised lignin streams. However, we note that many chemocatalytic upgrading strategies have a limited substrate scope since they require (i) volatile substrates for gas-phase conversion or (ii) substrates that are soluble in the applied solvent for liquid-phase conversion; HDO reactions are often performed in either water or alkane solvents, which both display a limited solubility for certain phenolic compounds. Moreover, the effect of poisons or oligomers in actual depolymerisation streams is largely unknown. As to the product side of the chemocatalytic funnel, an important consideration is that HDO processes almost exclusively aim at upgrading lignin depolymerisation streams to fuel-grade liquids (*e.g.* alkanes) instead of pure chemicals. The latter constitutes a rather new challenge in HDO research.

Similar to chemocatalytic funneling, the concept of using microbes to convert lignin-derived compounds to value-added products is still in its infancy. Significant research efforts are

needed to understand if this concept will be viable for biorefinery applications in the long term. Realistic biological processes generally require high titers, rates, and yields of products. In the context of biological funneling, this will likely hinge on the delivery of monomeric aromatic species or rapid biological depolymerisation of oligomeric lignin. Biocatalysis is most often performed in aqueous media which exhibit limited solubility for phenolic compounds, thereby putting constraints on how much carbon can get in the cell. In addition, not all compounds might go through the funnel fully, consequently leading to a lower conversion. Another important consideration is the possible detrimental effect of toxic aromatic substances present in the depolymerised lignin substrate. Certainly advances in lignin depolymerisation (Section 5) in some cases could link closely to downstream microbial conversions.

In conclusion, funneling is a promising but still new concept, and we strongly encourage future research in this direction. Though, it should be kept in mind that chemocatalytic and biocatalytic strategies each have their own (inherent) limitations. Paying attention to product selectivity already during lignin depolymerisation is therefore imperative, which underscores the connection between the three main biorefinery aspects (fractionation, depolymerisation, and upgrading).

7. Conclusion

During the last decade, there is a growing stimulus to better integrate lignin valorisation in current and future biorefinery schemes. While various (macromolecular) applications for lignin can be envisioned, lignin conversion to chemicals represents a promising opportunity, and is the focal point of this review. From a chemo-technological point of view, the success of a lignin-to-chemicals valorisation chain is determined by an interplay of three important biorefinery unit operations: (i) lignocellulose fractionation, (ii) lignin depolymerisation, and (iii) upgrading. For each of these aspects, diverse methodologies have been developed. Aligning these three interconnected transformations is of paramount importance to maximise the value harvested from lignin, and by extension, from the complete biomass.

The various lignocellulose fractionation methods can be split into two categories, namely (i) methods that target the extraction of lignin from the biomass, leaving behind a delignified carbohydrate pulp, and (ii) methods that intend to solubilise (or liquefy) the carbohydrate fractions, resulting in a solid lignin residue or precipitate. In either case, preventing structural degradation of lignin during fractionation – the key challenge for this subdomain – is important to preserve the reactivity towards subsequent depolymerisation. Newly developed fractionation methods therefore aim at (i) maximally retaining the native β-O-4 bonds in lignin and/or (ii) preventing the formation of recalcitrant carbon–carbon bonds once reactive intermediates are formed (see Fig. 13 for conceptual overview). Maximally retaining the native β-O-4 bonds in lignin can be accomplished by either active (chemical) stabilisation of the lignin structure (as in formaldehyde-assisted extraction), or passive β-O-4 preservation, by applying mild fractionation conditions or promoting the solubilisation of the biomass (*e.g.* GVL-assisted acid hydrolysis). Formation of recalcitrant carbon–carbon bonds can be prevented by chemically quenching reactive intermediates (as in RCF) or by physically removing them from the heating zone (as in flow-through processing). Hence, the successful implementation of these principles implies that the valorisation of lignin should be considered as a primary target of the biorefinery, right from the start, instead of a subordinate opportunity.

Efficient depolymerisation of the isolated lignins, *i.e.* the second stage of the lignin-to-chemicals valorisation chain, also requires that formation of stable carbon–carbon bonds (repolymerisation) is avoided. This can be accomplished by specifically aiming to prevent repolymerisation, whereas an alternative strategy is to enhance the reactivity of lignin towards depolymerisation, hereby increasing the rate of depolymerisation relative to the rate of repolymerisation (Fig. 23). As in biomass fractionation, repolymerisation of reactive intermediates can be prevented by chemical quenching (*e.g.* through reductive processing or ACD combined with acetal formation or decarbonylation) or physical removal from the reaction zone. Enhancing the reactivity of the β-O-4 ether bonds has been demonstrated by both benzylic alcohol oxidation and methylation.

In spite of the innovations made in the field of biomass fractionation, industrial implementation will require significant additional research efforts. At least in the short term, the paper industry will continue to produce high volumes of lignin according to well-established traditional methods which inevitably induce severe lignin degradation. In particular, kraft pulping is likely to remain the predominant pulping process for the years to come. Hence, valorisation of kraft and other degraded lignins – as long as they are not consumed to generate required local energy – remains a difficult but important objective to the biorefinery. Depolymerisation methods that are able to (selectively) cleave carbon–carbon bonds are probably most suitable to turn these recalcitrant substrates into chemicals. Such methods often require harsh processing conditions, which in turn promote the occurrence of repolymerisation (lowering the monomer yield) and unwanted secondary reactions (lowering the monomer selectivity). Hence, degraded lignins constitute a challenging feedstock for the production of chemicals, which poses the question whether or not these substrates might better be utilised for macromolecular applications (*i.e.* lignin-to-materials).

As complex product mixtures are frequently obtained from lignin depolymerisation, upgrading these mixtures to targeted chemicals is the last link in the lignin-to-chemicals valorisation chain. An encouraging concept within this third subdomain is funneling. The goal of this concept is to transform (*i.e.* funnel) broad and diverse product mixtures to a smaller set of central intermediates, which in turn can be converted to desired chemicals. Hence, funneling can serve as a tool to bridge the gap between raw product soups obtained from lignin depolymerisation, and the application of specific chemicals and fuels. This tool can be translated into chemocatalytic methods (*e.g.* defunctionalisation *via* HDO) as well as into biological approaches by engineering

nature's inherent metabolic funnels. Several upgrading routes adopting this funneling principle have been demonstrated, ranging from the production of fuel compounds over drop-in chemicals to innovative polymer building blocks. Keeping in mind that funneling also has its limitations, it remains important to pay attention to the selectivity of lignin depolymerisation processes.

Finally, to guide and improve future research on fractionation/depolymerisation, we would like to point out a few critical remarks. As learnt from the above overview, the structure and reactivity of isolated lignins is governed by (i) the biomass type, (ii) the fractionation method, and (iii) the harshness of the fractionation method (which in turn determines the isolated lignin yield or purity). Therefore, fair comparisons of structural characteristics can only be made by decoupling these three determining aspects from one another. For example, in order to make an unambiguous assessment of different fractionation methods, the biomass source should be identical, and the isolated lignin yields (or purity) should at least be similar. Likewise, with respect to lignin depolymerisation, the effectiveness strongly depends on (i) the depolymerisation method as well as on (ii) the structural characteristic of the lignin substrate. A clear comparison of different depolymerisation methods requires that the lignin substrate is the same, and *vice versa*.

When these prerequisites are fulfilled, fractionation and depolymerisation methods can be evaluated over different studies. However, an additional hurdle is the large variation in analytical procedures that exist between studies, leading to different outcomes. Hence, to facilitate a fair and trustworthy assessment of the various fractionation and depolymerisation methods, comparative studies that keep in mind the proposed criteria and apply the same set of analytical methods can provide valuable insight.

As last, we would like to stimulate the lignin research community to take into account the entire (native) lignin fraction in future fractionation and lignin depolymerisation studies. An optimal valorisation of lignin into chemicals requires an optimal use of the entire lignin portion in the feedstock, which thus necessitates the combination of an effective (high yield) lignin isolation and depolymerisation step.

Abbreviations

AAP	Anhydrous ammonia pretreatment
AFEX	Ammonia fiber explosion
ACD	Acid-catalysed depolymerisation
APL	Alkaline pretreatment liquor
AQ	Anthraquinone
ARP	Ammonia recycled percolation
ASL	Acid-soluble lignin
BCD	Base-catalysed depolymerisation
BuOH	Butanol
CAH	Concentrated acid hydrolysis
CEL	Cellulolytic enzyme lignin
CFP	Catalytic fast pyrolysis
DAH	Dilute acid hydrolysis
DAP	Dilute acid pretreatment
DL	Depolymerised lignin
DMR	Deacetylating and mechanical refining
EAP	Extractive ammonia pretreatment
ECCL	Early-stage catalytic conversion of lignin
EMAL	Enzymatic mild acidolysis lignin
EtGly	Ethylene glycol
EtOH	Ethanol
FA	Formaldehyde
FT	Flow-through
FP	Fast pyrolysis
G	Guaiacyl or guaiacol
GC	Gas chromatography
GPC	Gel permeation chromatography
GVL	γ-Valerolactone
H	*para*-Hydroxyphenyl
HDO	Hydrodeoxygenation
HPA	Heteropolyacid
HSQC	Heteronuclear single quantum coherence
HTC	Hydroxytalcite
HWP	Hot water pretreatment
im	Imidazolium
iPrOH	Isopropanol
IL	Ionic liquid
LCC	Lignin–carbohydrate complex
LP	Lignin precipitate
LR	Lignin residue
MeOH	Methanol
MeTHF	Methyl tetrahydrofuran
MIBK	Methy isobutyl ketone
M(OTf)	Metal triflate
MWL	Milled wood lignin
NMR	Nuclear magnetic resonance (spectroscopy)
NP(s)	Nanoparticle(s)
MW	Molecular weight
PMO	Porous metal oxide
RCF	Reductive catalytic fractionation
RT	Room temperature
S	Syringyl or syringol
SEP	Steam explosion pretreatment
THF	Tetrahydrofuran

Conflicts of interest

There are no conflicts to declare.

Acknowledgements

This work was performed in the framework of Catalisti (formerly FISCH)-SBO project ARBOREF and EU Interreg Vlaanderen-Nederland project BIO-HArT. W. S., T. R., and S. V. d. B. acknowledge the Research Foundation Flanders (FWO Vlaanderen) for (post-)doctoral fellowships. S.-F. K. acknowledges funding from Catalisti-SBO project ARBOREF. G. T. B. acknowledges

funding from the US Department of Energy Biotechnologies Office *via* Contract No. DE-AC36-08GO28308 to the National Renewable Energy Laboratory. G. T. B. also acknowledges funding from the Direct Catalytic Conversion of Biomass to Biofuels (C3Bio), an Energy Frontier Research Center funded by the U.S. Department of Energy, Office of Science, Office of Basic Energy Sciences, Award Number DE-SC0000997 and from the BioEnergy Science Center, a U.S. Department of Energy Bioenergy Research Center supported by the Office of Biological and Environmental Research in the DOE Office of Science.

Notes and references

1 A. J. Ragauskas, C. K. Williams, B. H. Davison, G. Britovsek, J. Cairney, C. A. Eckert, W. J. Frederick, J. P. Hallett, D. J. Leak and C. L. Liotta, *Science*, 2006, **311**, 484–489.
2 M. I. Hoffert, K. Caldeira, G. Benford, D. R. Criswell, C. Green, H. Herzog, A. K. Jain, H. S. Kheshgi, K. S. Lackner, J. S. Lewis, H. D. Lightfoot, W. Manheimer, J. C. Mankins, M. E. Mauel, L. J. Perkins, M. E. Schlesinger, T. Volk and T. M. L. Wigley, *Science*, 2002, **298**, 981–987.
3 B. H. Davison, J. Parks, M. F. Davis and B. S. Donohoe, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, ed. C. E. Wyman, John Wiley & Sons, Ltd, 2013, pp. 23–38.
4 J. E. Bidlack and W. V. Dashek, *Plant Cells and their Organelles*, John Wiley & Sons, Ltd, 2017, pp. 209–238.
5 A.-M. Boudet, in *The plant cell wall*, ed. J. K. C. Rose, Blackwell Publishing Ltd, Oxford, 2003, vol. 8, pp. 155–182.
6 M. E. Himmel, S.-Y. Ding, D. K. Johnson, W. S. Adney, M. R. Nimlos, J. W. Brady and T. D. Foust, *Science*, 2007, **315**, 804–807.
7 J. D. DeMartini, S. Pattathil, J. S. Miller, H. Li, M. G. Hahn and C. E. Wyman, *Energy Environ. Sci.*, 2013, **6**, 898–909.
8 M. Ragnar, G. Henriksson, M. E. Lindström, M. Wimby, J. Blechschmidt and S. Heinemann, in *Ullmann's Encyclopedia of Industrial Chemistry*, Wiley-VCH Verlag GmbH & Co. KGaA, 2000.
9 B. Hahn-Hägerdal, M. Galbe, M. F. Gorwa-Grauslund, G. Lidén and G. Zacchi, *Trends Biotechnol.*, 2006, **24**, 549–556.
10 A. Limayem and S. C. Ricke, *Prog. Energy Combust. Sci.*, 2012, **38**, 449–467.
11 A. J. Ragauskas, G. T. Beckham, M. J. Biddy, R. Chandra, F. Chen, M. F. Davis, B. H. Davison, R. A. Dixon, P. Gilna, M. Keller, P. Langan, A. K. Naskar, J. N. Saddler, T. J. Tschaplinski, G. A. Tuskan and C. E. Wyman, *Science*, 2014, **344**, 709.
12 C. O. Tuck, E. Perez, I. T. Horvath, R. A. Sheldon and M. Poliakoff, *Science*, 2012, **337**, 695–699.
13 J. E. Holladay, J. F. White, J. J. Bozell and D. Johnson, *Top Value-Added Chemicals from Biomass – Volume II—Results of Screening for Potential Candidates from Biorefinery Lignin, Report PNNL-16983*, Pacific Northwest National Laboratory (PNNL), Richland, WA (US), 2007.
14 R. Davis, L. Tao, E. Tan, M. Biddy, G. Beckham, C. Scarlata, J. Jacobson, K. Cafferty, J. Ross and J. Lukas, *Process design and economics for the conversion of lignocellulosic biomass to hydrocarbons: dilute-acid and enzymatic deconstruction of biomass to sugars and biological conversion of sugars to hydrocarbons*, Report NREL/TP-5100-60223, National Renewable Energy Laboratory (NREL), Goldon, CO (US), 2013.
15 B. M. Upton and A. M. Kasko, *Chem. Rev.*, 2016, **116**, 2275–2306.
16 S. Sen, S. Patil and D. S. Argyropoulos, *Green Chem.*, 2015, **17**, 4862–4887.
17 W.-J. Liu, H. Jiang and H.-Q. Yu, *Green Chem.*, 2015, **17**, 4888–4907.
18 D. Esposito and M. Antonietti, *Chem. Soc. Rev.*, 2015, **44**, 5821–5835.
19 H. Chung and N. R. Washburn, *Green Mater.*, 2013, **1**, 137–160.
20 M. Graglia, N. Kanna and D. Esposito, *ChemBioEng Rev.*, 2015, 377–392.
21 R. Rinaldi, R. Jastrzebski, M. T. Clough, J. Ralph, M. Kennema, P. C. A. Bruijnincx and B. M. Weckhuysen, *Angew. Chem., Int. Ed.*, 2016, **55**, 8164–8215.
22 J. J. Bozell, in *Selective Catalysis for Renewable Feedstocks and Chemicals*, ed. K. M. Nicholas, 2014, vol. 353, pp. 229–255.
23 C. Li, X. Zhao, A. Wang, G. W. Huber and T. Zhang, *Chem. Rev.*, 2015, **115**, 11559–11624.
24 J. Zakzeski, P. C. A. Bruijnincx, A. L. Jongerius and B. M. Weckhuysen, *Chem. Rev.*, 2010, **110**, 3552–3599.
25 I. Delidovich, K. Leonhard and R. Palkovits, *Energy Environ. Sci.*, 2014, **7**, 2803–2830.
26 I. Delidovich, P. J. C. Hausoul, L. Deng, R. Pfützenreuter, M. Rose and R. Palkovits, *Chem. Rev.*, 2016, **116**, 1540–1599.
27 S. Van de Vyver, J. Geboers, P. A. Jacobs and B. F. Sels, *ChemCatChem*, 2011, **3**, 82–94.
28 A. Corma, S. Iborra and A. Velty, *Chem. Rev.*, 2007, **107**, 2411–2502.
29 G. W. Huber, S. Iborra and A. Corma, *Chem. Rev.*, 2006, **106**, 4044–4098.
30 D. Klemm, B. Heublein, H. P. Fink and A. Bohn, *Angew. Chem., Int. Ed.*, 2005, **44**, 3358–3393.
31 H. Chen, *Biotechnology of Lignocellulose: Theory and Practice*, Springer, Netherlands, Dordrecht, 2014, pp. 25–71.
32 T. Heinze, in *Cellulose Chemistry and Properties: Fibers, Nanocelluloses and Advanced Materials*, ed. J. O. Rojas, Springer International Publishing, Cham, 2016, pp. 1–52.
33 P. Harmsen, W. Huijgen, L. Bermudez and R. Bakker, *Literature review of physical and chemical pretreatment processes for lignocellulosic biomass*, 2010.
34 Y. Nishiyama, P. Langan and H. Chanzy, *J. Am. Chem. Soc.*, 2002, **124**, 9074–9082.
35 Y. Nishiyama, J. Sugiyama, H. Chanzy and P. Langan, *J. Am. Chem. Soc.*, 2003, **125**, 14300–14306.
36 F. M. Gírio, C. Fonseca, F. Carvalheiro, L. C. Duarte, S. Marques and R. Bogel-Łukasik, *Bioresour. Technol.*, 2010, **101**, 4775–4800.
37 J. Puls, *Macromol. Symp.*, 1997, **120**, 183–196.

38 K. C. B. Wilkie, in *Advances in Carbohydrate Chemistry and Biochemistry*, ed. R. S. Tipson and H. Derek, Academic Press, 1979, vol. 36, pp. 215–264.
39 R. L. Silveira, S. R. Stoyanov, S. Gusarov, M. S. Skaf and A. Kovalenko, *J. Am. Chem. Soc.*, 2013, **135**, 19048–19051.
40 Y. Pu, D. Zhang, P. M. Singh and A. J. Ragauskas, *Biofuels, Bioprod. Biorefin.*, 2008, **2**, 58–73.
41 N. S. Thompson, in *Kirk-Othmer Encyclopedia of Chemical Technology*, John Wiley & Sons, Inc., 2000.
42 M. B. G. Latarullo, E. Q. P. Tavares, G. P. Maldonado, D. C. C. Leite and M. S. Buckeridge, *Front. Plant Sci.*, 2016, **7**, 1401.
43 M. A. Atmodjo, Z. Hao and D. Mohnen, *Annu. Rev. Plant Biol.*, 2013, **64**, 747–779.
44 W. Boerjan, J. Ralph and M. Baucher, *Annu. Rev. Plant Biol.*, 2003, **54**, 519–546.
45 J. Ralph, K. Lundquist, G. Brunow, F. Lu, H. Kim, P. Schatz, J. Marita, R. Hatfield, S. Ralph, J. Christensen and W. Boerjan, *Phytochem. Rev.*, 2004, **3**, 29–60.
46 R. Vanholme, B. Demedts, K. Morreel, J. Ralph and W. Boerjan, *Plant Physiol.*, 2010, **153**, 895–905.
47 K. E. Achyuthan, A. M. Achyuthan, P. D. Adams, S. M. Dirk, J. C. Harper, B. A. Simmons and A. K. Singh, *Molecules*, 2010, **15**, 8641.
48 B. Saake and R. Lehnen, in *Ullmann's Encyclopedia of Industrial Chemistry*, Wiley-VCH Verlag GmbH & Co. KGaA, 2000.
49 J. Barros, J. C. Serrani-Yarce, F. Chen, D. Baxter, B. J. Venables and R. A. Dixon, *Nat. Plants*, 2016, **2**, 16050.
50 H. A. Maeda, *Nat. Plants*, 2016, **2**, 16080.
51 R. Vanholme, K. Morreel, C. Darrah, P. Oyarce, J. H. Grabber, J. Ralph and W. Boerjan, *New Phytol.*, 2012, **196**, 978–1000.
52 J. Ralph, *Phytochem. Rev.*, 2010, **9**, 65–83.
53 D. C. C. Smith, *J. Chem. Soc.*, 1955, 2347–2351.
54 F. Lu, J. Ralph, K. Morreel, E. Messens and W. Boerjan, *Org. Biomol. Chem.*, 2004, **2**, 2888–2890.
55 K. Morreel, J. Ralph, H. Kim, F. C. Lu, G. Goeminne, S. Ralph, E. Messens and W. Boerjan, *Plant Physiol.*, 2004, **136**, 3537–3549.
56 J. C. del Río, J. Rencoret, P. Prinsen, Á. T. Martínez, J. Ralph and A. Gutiérrez, *J. Agric. Food Chem.*, 2012, **60**, 5922–5935.
57 W. Lan, F. Lu, M. Regner, Y. Zhu, J. Rencoret, S. A. Ralph, U. I. Zakai, K. Morreel, W. Boerjan and J. Ralph, *Plant Physiol.*, 2015, **167**, 1284–1295.
58 M. Li, Y. Pu, C. G. Yoo and A. J. Ragauskas, *Green Chem.*, 2016, **18**, 1439–1454.
59 W. Lan, K. Morreel, F. Lu, J. Rencoret, J. C. del Río, W. Voorend, W. Vermerris, W. A. Boerjan and J. Ralph, *Plant Physiol.*, 2016, **171**, 810–820.
60 J. C. Del Río, G. Marques, J. Rencoret, Á. T. Martínez and A. Gutiérrez, *J. Agric. Food Chem.*, 2007, **55**, 5461–5468.
61 J. Ralph, *J. Nat. Prod.*, 1996, **59**, 341–342.
62 Y. Mottiar, R. Vanholme, W. Boerjan, J. Ralph and S. D. Mansfield, *Curr. Opin. Biotechnol.*, 2016, **37**, 190–200.
63 R. Vanholme, K. Morreel, J. Ralph and W. Boerjan, *Curr. Opin. Plant Biol.*, 2008, **11**, 278–285.
64 M. V. Galkin and J. S. M. Samec, *ChemSusChem*, 2016, **9**, 1544–1558.
65 R. Parthasarathi, R. A. Romero, A. Redondo and S. Gnanakaran, *J. Phys. Lett.*, 2011, **2**, 2660–2666.
66 D. D. Laskar, B. Yang, H. M. Wang and J. Lee, *Biofuels, Bioprod. Biorefin.*, 2013, **7**, 602–626.
67 A. Tolbert, H. Akinosho, R. Khunsupat, A. K. Naskar and A. J. Ragauskas, *Biofuels, Bioprod. Biorefin.*, 2014, **8**, 836–856.
68 S. P. Chundawat, G. T. Beckham, M. E. Himmel and B. E. Dale, *Annu. Rev. Chem. Biomol. Eng.*, 2011, **2**, 121–145.
69 T.-Q. Yuan, S.-N. Sun, F. Xu and R.-C. Sun, *J. Agric. Food Chem.*, 2011, **59**, 10604–10614.
70 M. Balakshin, E. Capanema, H. Gracz, H.-m. Chang and H. Jameel, *Planta*, 2011, **233**, 1097–1110.
71 H. Wang, J. Male and Y. Wang, *ACS Catal.*, 2013, **3**, 1047–1070.
72 G. Gellerstedt and G. Henriksson, *Lignins: major sources, structure and properties*, Amsterdam, The Netherlands, Elsevier, 2008.
73 P. Azadi, O. R. Inderwildi, R. Farnood and D. A. King, *Renewable Sustainable Energy Rev.*, 2013, **21**, 506–523.
74 A.-M. Boudet, *Plant Physiol. Biochem.*, 2000, **38**, 81–96.
75 S. Constant, H. L. J. Wienk, A. E. Frissen, P. d. Peinder, R. Boelens, D. S. van Es, R. J. H. Grisel, B. M. Weckhuysen, W. J. J. Huijgen, R. J. A. Gosselink and P. C. A. Bruijnincx, *Green Chem.*, 2016, **18**, 2651–2665.
76 T. Aro and P. Fatehi, *ChemSusChem*, 2017, **10**, 1861–1877.
77 F. G. Calvo-Flores and J. A. Dobado, *ChemSusChem*, 2010, **3**, 1227–1235.
78 W. O. Doherty, P. Mousavioun and C. M. Fellows, *Ind. Crops Prod.*, 2011, **33**, 259–276.
79 S. Laurichesse and L. Avérous, *Prog. Polym. Sci.*, 2014, **39**, 1266–1290.
80 F. G. Calvo-Flores, J. A. Dobado, J. Isac-García and F. J. Martín-MartíNez, in *Lignin and Lignans as Renewable Raw Materials: Chemistry, Technology and Applications*, John Wiley & Sons, Ltd, 2015, pp. 247–288.
81 G. Gellerstedt, in *Pulping Chemistry and Technology*, ed. M. Ek, G. Gellerstedt and G. Henriksson, 2009, vol. 2, pp. 91–120.
82 J. Gierer, *Wood Sci. Technol.*, 1980, **14**, 241–266.
83 E. Sjöström, in *Wood Chemistry*, Academic Press, San Diego, 2nd edn, 1993, pp. 114–164.
84 R. S. Ramirez, M. Holtzapple and N. Piamonte, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, ed. C. E. Wyman, John Wiley & Sons, Ltd, 2013, pp. 145–167.
85 J. Gierer, *Wood Sci. Technol.*, 1985, **19**, 289–312.
86 J. Gierer, *Holzforschung*, 1982, **36**, 43–51.
87 J. Gierer, I. Noren and S. Wannstrom, *Holzforschung*, 1987, **41**, 79–82.
88 D. Dimmel and G. Gellerstedt, in *Lignin and Lignans*, CRC Press, 2010, pp. 349–391.
89 F. S. Chakar and A. J. Ragauskas, *Ind. Crops Prod.*, 2004, **20**, 131–141.
90 Y. Pu, F. Hu, F. Huang, B. H. Davison and A. J. Ragauskas, *Biotechnol. Biofuels*, 2013, **6**, 15.

91 Y. Pu, F. Hu, F. Huang and A. J. Ragauskas, *BioEnergy Res.*, 2015, **8**, 992–1003.
92 M. R. Sturgeon, S. Kim, K. Lawrence, R. S. Paton, S. C. Chmely, M. Nimlos, T. D. Foust and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2014, **2**, 472–485.
93 T. Imai, T. Yokoyama and Y. Matsumoto, *J. Wood Sci.*, 2011, **57**, 219–225.
94 T. Yokoyama, *J. Wood Chem. Technol.*, 2015, **35**, 27–42.
95 O. Karlsson, K. Lundquist, S. Meuller and K. Westlid, *Acta Chem. Scand., Ser. B*, 1988, **42**, 48–51.
96 K. Lundquist and R. Lundgren, *Acta Chem. Scand.*, 1972, **26**, 2005–2023.
97 P. J. Deuss, M. Scott, F. Tran, N. J. Westwood, J. G. de Vries and K. Barta, *J. Am. Chem. Soc.*, 2015, **137**, 7456–7467.
98 E. Adler, *Wood Sci. Technol.*, 1977, **11**, 169–218.
99 M. Kulka and H. Hibbert, *J. Am. Chem. Soc.*, 1943, **65**, 1180–1185.
100 A. Cramer, M. Hunter and H. Hibbert, *J. Am. Chem. Soc.*, 1939, **61**, 509–516.
101 E. West, A. MacInnes and H. Hibbert, *J. Am. Chem. Soc.*, 1943, **65**, 1187–1192.
102 P. Sannigrahi, A. J. Ragauskas and S. J. Miller, *Energy Fuels*, 2009, **24**, 683–689.
103 M. Zaheer and R. Kempe, *ACS Catal.*, 2015, **5**, 1675–1684.
104 M. V. Galkin, S. Sawadjoon, V. Rohde, M. Dawange and J. S. Samec, *ChemCatChem*, 2014, **6**, 179–184.
105 J. Lu, M. Wang, X. Zhang, A. Heyden and F. Wang, *ACS Catal.*, 2016, **6**, 5589–5598.
106 T. H. Parsell, B. C. Owen, I. Klein, T. M. Jarrell, C. L. Marcum, L. J. Haupert, L. M. Amundson, H. I. Kenttämaa, F. Ribeiro and J. T. Miller, *Chem. Sci.*, 2013, **4**, 806–813.
107 H. Guo, B. Zhang, C. Li, C. Peng, T. Dai, H. Xie, A. Wang and T. Zhang, *ChemSusChem*, 2016, **9**, 3220–3229.
108 R. Ma, Y. Xu and X. Zhang, *ChemSusChem*, 2015, **8**, 24–51.
109 J. Gierer, *Wood Sci. Technol.*, 1986, **20**, 1–33.
110 J. Gierer, F. Imsgard and I. Norén, *Acta Chem. Scand.*, 1977, **B31**, 561–572.
111 V. E. Tarabanko, Y. V. Hendogina, D. V. Petuhov and E. P. Pervishina, *React. Kinet. Catal. Lett.*, 2000, **69**, 361–368.
112 V. E. Tarabanko and D. V. Petukhov, *Chem. Sustainable Dev.*, 2003, **11**, 655–667.
113 V. E. Tarabanko, D. V. Petukhov and G. E. Selyutin, *Kinet. Catal.*, 2004, **45**, 569–577.
114 C. Fargues, Á. Mathias, J. Silva and A. Rodrigues, *Chem. Eng. Technol.*, 1996, **19**, 127–136.
115 P. R. Patwardhan, R. C. Brown and B. H. Shanks, *ChemSusChem*, 2011, **4**, 1629–1636.
116 W. Mu, H. Ben, A. Ragauskas and Y. Deng, *BioEnergy Res.*, 2013, **6**, 1183–1204.
117 S. Wang, G. Dai, H. Yang and Z. Luo, *Prog. Energy Combust. Sci.*, 2017, **62**, 33–86.
118 H. Kawamoto, *J. Wood Sci.*, 2017, **63**, 117–132.
119 B. Holmelid, M. Kleinert and T. Barth, *J. Anal. Appl. Pyrolysis*, 2012, **98**, 37–44.
120 J.-Y. Liu, S.-B. Wu and R. Lou, *BioResources*, 2011, **6**, 1079–1093.
121 P. F. Britt, A. C. Buchanan, M. J. Cooney and D. R. Martineau, *J. Inorg. Chem.*, 2000, **65**, 1376–1389.
122 P. F. Britt, M. K. Kidder and A. C. Buchanan, *Energy Fuels*, 2007, **21**, 3102–3108.
123 S. Chu, A. V. Subrahmanyam and G. W. Huber, *Green Chem.*, 2013, **15**, 125–136.
124 M. T. Klein and P. S. Virk, *Ind. Eng. Chem. Fundam.*, 1983, **22**, 35–45.
125 M. W. Jarvis, J. W. Daily, H.-H. Carstensen, A. M. Dean, S. Sharma, D. C. Dayton, D. J. Robichaud and M. R. Nimlos, *J. Phys. Chem. A*, 2011, **115**, 428–438.
126 X. Huang, C. Liu, J. Huang and H. Li, *Comput. Theor. Chem.*, 2011, **976**, 51–59.
127 T. Elder and A. Beste, *Energy Fuels*, 2014, **28**, 5229–5235.
128 Y. S. Choi, R. Singh, J. Zhang, G. Balasubramanian, M. R. Sturgeon, R. Katahira, G. Chupka, G. T. Beckham and B. H. Shanks, *Green Chem.*, 2016, **18**, 1762–1773.
129 L. D. Dellon, A. J. Yanez, W. Li, R. Mabon and L. J. Broadbelt, *Energy Fuels*, 2017, **31**, 8263–8274.
130 A. J. Yanez, W. Li, R. Mabon and L. J. Broadbelt, *Energy Fuels*, 2016, **30**, 5835–5845.
131 R. Vinu and L. J. Broadbelt, *Energy Environ. Sci.*, 2012, **5**, 9808–9826.
132 To clarify the difference between a lignin precipitate (LP) and lignin residue (LR): LP is obtained from fractionation methods that extract and solubilise lignin, followed by down-stream precipitation from the solution (*e.g.* kraft lignin). LR is obtained from processes that solubilise/liquefy carbohydrates, without solubilising lignin. Hence, the lignin fraction is recovered as a residual solid (*e.g.* Klason lignin).
133 R. Patt, O. Kordsachia, R. Süttinger, Y. Ohtani, J. F. Hoesch, P. Ehrler, R. Eichinger, H. Holik, U. Hamm and M. E. Rohmann, *Ullmann's Encyclopedia of Industrial Chemistry*, 2000.
134 F. G. Calvo-Flores, J. A. Dobado, J. Isac-García and F. J. Martín-MartíNez, in *Lignin and Lignans as Renewable Raw Materials: Chemistry*, Technology and Applications, John Wiley & Sons, Ltd, 2015, pp. 113–144.
135 C. S. Lancefield, G. M. M. Rashid, F. Bouxin, A. Wasak, W.-C. Tu, J. Hallett, S. Zein, J. Rodríguez, S. D. Jackson, N. J. Westwood and T. D. H. Bugg, *ACS Sustainable Chem. Eng.*, 2016, **4**, 6921–6930.
136 P. Prinsen, J. Rencoret, A. Gutiérrez, T. Liitiä, T. Tamminen, J. L. Colodette, M. Á. Berbis, J. Jiménez-Barbero, Á. T. Martínez and J. C. del Río, *Ind. Eng. Chem. Res.*, 2013, **52**, 15702–15712.
137 J.-P. Lange, *Angew. Chem., Int. Ed.*, 2015, **54**, 13186–13197.
138 J. Lora, in *Monomers, Polymers and Composites from Renewable Resources*, ed. M. N. B. Gandini and M. N. Belgacem, Elsevier, Amsterdam, 2008, pp. 225–241.
139 A. P. Marques, D. V. Evtuguin, S. Magina, F. M. L. Amado and A. Prates, *J. Wood Chem. Technol.*, 2009, **29**, 337–357.
140 S. Magina, P. Marques Ana and V. Evtuguin Dmitry, *Holzforschung*, 2015, **69**, 513.
141 D. R. Dimmel, *J. Wood Chem. Technol.*, 1985, **5**, 1–14.

142 R. Francis, S. J. Shin, S. Omori, T. Amidon and T. Blain, *J. Wood Chem. Technol.*, 2006, **26**, 141–152.
143 F. P. Bouxin, A. McVeigh, F. Tran, N. J. Westwood, M. C. Jarvis and S. D. Jackson, *Green Chem.*, 2015, **17**, 1235–1242.
144 E. M. Karp, C. T. Nimlos, S. Deutch, D. Salvachúa, R. M. Cywar and G. T. Beckham, *Green Chem.*, 2016, **18**, 4750–4760.
145 J. G. Linger, D. R. Vardon, M. T. Guarnieri, E. M. Karp, G. B. Hunsinger, M. A. Franden, C. W. Johnson, G. Chupka, T. J. Strathmann, P. T. Pienkos and G. T. Beckham, *Proc. Natl. Acad. Sci. U. S. A.*, 2014, **111**, 12013–12018.
146 D. R. Vardon, M. A. Franden, C. W. Johnson, E. M. Karp, M. T. Guarnieri, J. G. Linger, M. J. Salm, T. J. Strathmann and G. T. Beckham, *Energy Environ. Sci.*, 2015, **8**, 617–628.
147 E. M. Karp, M. G. Resch, B. S. Donohoe, P. N. Ciesielski, M. H. O'Brien, J. E. Nill, A. Mittal, M. J. Biddy and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2015, **3**, 1479–1491.
148 V. Chang, M. Nagwani and M. Holtzapple, *Appl. Biochem. Biotechnol.*, 1998, **74**, 135–159.
149 W. E. Kaar and M. T. Holtzapple, *Biomass Bioenergy*, 2000, **18**, 189–199.
150 S. Kim and M. T. Holtzapple, *Bioresour. Technol.*, 2005, **96**, 1994–2006.
151 E. M. Karp, B. S. Donohoe, M. H. O'Brien, P. N. Ciesielski, A. Mittal, M. J. Biddy and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2014, **2**, 1481–1491.
152 Z. Strassberger, P. Prinsen, F. v. d. Klis, D. S. v. Es, S. Tanase and G. Rothenberg, *Green Chem.*, 2015, **17**, 325–334.
153 P. Prinsen, A. Narani and G. Rothenberg, *ChemSusChem*, 2017, **10**, 1022–1032.
154 S. P. S. Chundawat, B. S. Donohoe, L. da Costa Sousa, T. Elder, U. P. Agarwal, F. Lu, J. Ralph, M. E. Himmel, V. Balan and B. E. Dale, *Energy Environ. Sci.*, 2011, **4**, 973–984.
155 L. da Costa Sousa, M. Jin, S. P. S. Chundawat, V. Bokade, X. Tang, A. Azarpira, F. Lu, U. Avci, J. Humpula, N. Uppugundla, C. Gunawan, S. Pattathil, A. M. Cheh, N. Kothari, R. Kumar, J. Ralph, M. G. Hahn, C. E. Wyman, S. Singh, B. A. Simmons, B. E. Dale and V. Balan, *Energy Environ. Sci.*, 2016, **9**, 1215–1223.
156 A. Mittal, R. Katahira, B. S. Donohoe, S. Pattathil, S. Kandemkavil, M. L. Reed, M. J. Biddy and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2017, **5**, 2544–2561.
157 V. Balan, B. Bals, S. P. S. Chundawat, D. Marshall and B. E. Dale, in *Biofuels: Methods and Protocols*, ed. R. J. Mielenz, Humana Press, Totowa, NJ, 2009, pp. 61–77.
158 S. P. S. Chundawat, R. Vismeh, L. N. Sharma, J. F. Humpula, L. da Costa Sousa, C. K. Chambliss, A. D. Jones, V. Balan and B. E. Dale, *Bioresour. Technol.*, 2010, **101**, 8429–8438.
159 J. S. Kim, Y. Y. Lee and T. H. Kim, *Bioresour. Technol.*, 2016, **199**, 42–48.
160 S. P. S. Chundawat, B. Bals, T. Campbell, L. Sousa, D. Gao, M. Jin, P. Eranki, R. Garlock, F. Teymouri, V. Balan and B. E. Dale, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, John Wiley & Sons, Ltd, 2013, pp. 169–200.
161 S. Singh, G. Cheng, N. Sathitsuksanoh, D. Wu, P. Varanasi, A. George, V. Balan, X. Gao, R. Kumar, B. E. Dale, C. E. Wyman and B. A. Simmons, *Front. Energy Res.*, 2015, **2**, 62.
162 L. da Costa Sousa, M. Foston, V. Bokade, A. Azarpira, F. Lu, A. J. Ragauskas, J. Ralph, B. Dale and V. Balan, *Green Chem.*, 2016, **18**, 4205–4215.
163 M. Wada, H. Chanzy, Y. Nishiyama and P. Langan, *Macromolecules*, 2004, **37**, 8548–8555.
164 S. P. S. Chundawat, G. Bellesia, N. Uppugundla, L. da Costa Sousa, D. Gao, A. M. Cheh, U. P. Agarwal, C. M. Bianchetti, G. N. Phillips, P. Langan, V. Balan, S. Gnanakaran and B. E. Dale, *J. Am. Chem. Soc.*, 2011, **133**, 11163–11174.
165 D. Gao, S. P. Chundawat, A. Sethi, V. Balan, S. Gnanakaran and B. E. Dale, *Proc. Natl. Acad. Sci. U. S. A.*, 2013, **110**, 10922–10927.
166 T. H. Kim, J. S. Kim, C. Sunwoo and Y. Y. Lee, *Bioresour. Technol.*, 2003, **90**, 39–47.
167 T. H. Kim and Y. Y. Lee, *Bioresour. Technol.*, 2005, **96**, 2007–2013.
168 H. H. Yoon, Z. W. Wu and Y. Y. Lee, *Appl. Biochem. Biotechnol.*, 1995, **51**, 5–19.
169 P. V. Iyer, Z.-W. Wu, S. B. Kim and Y. Y. Lee, in *Seventeenth Symposium on Biotechnology for Fuels and Chemicals: Presented as Volumes 57 and 58 of Applied Biochemistry and Biotechnology*, ed. C. E. Wyman and B. H. Davison, Humana Press, Totowa, NJ, 1996, pp. 121–132.
170 B. Yang and C. E. Wyman, *Biofuels, Bioprod. Biorefin.*, 2008, **2**, 26–40.
171 F. P. Bouxin, S. David Jackson and M. C. Jarvis, *Bioresour. Technol.*, 2014, **162**, 236–242.
172 C. Liu and C. E. Wyman, *Ind. Eng. Chem. Res.*, 2004, **43**, 2781–2788.
173 C. Liu and C. E. Wyman, *Ind. Eng. Chem. Res.*, 2003, **42**, 5409–5416.
174 S. Bhagia, H. Li, X. Gao, R. Kumar and C. E. Wyman, *Biotechnol. Biofuels*, 2016, **9**, 245.
175 B. S. Donohoe, S. R. Decker, M. P. Tucker, M. E. Himmel and T. B. Vinzant, *Biotechnol. Bioeng.*, 2008, **101**, 913–925.
176 B. Yang and C. E. Wyman, *Biotechnol. Bioeng.*, 2004, **86**, 88–98.
177 To the best of our knowledge, no direct information was found on the beta-O-4 content of lignin obtained from FT-DAP/HWP. Nevertheless, studies on the depolymerisation of lignin obtained from flow-through dilute acid hydrolysis indicate that these type of lignins have a high reactivity, which suggests that beta-O-4 linkages are at least partly preserved. Because, FT-DAP/HWP is less severe compared to complete hydrolysis, it can be assumed that these delignification methods also partially preserve the beta-O-4 linkages.
178 H. Wang, H. Ben, H. Ruan, L. Zhang, Y. Pu, M. Feng, A. J. Ragauskas and B. Yang, *ACS Sustainable Chem. Eng.*, 2017, **5**, 1824–1830.
179 Q. Xiang and Y. Y. Lee, *Appl. Biochem. Biotechnol.*, 2000, **84**, 153–162.
180 Q. Xiang and Y. Y. Lee, *Appl. Biochem. Biotechnol.*, 2001, **91**, 71–80.

181 H. Wang, H. Ruan, M. Feng, Y. Qin, H. Job, L. Luo, C. Wang, M. H. Engelhard, E. Kuhn and X. Chen, *ChemSusChem*, 2017, **10**, 1846–1856.
182 L. Yan, R. Ma, L. Li and J. Fu, *Chem. Eng. Technol.*, 2016, **39**, 1759–1770.
183 N. S. Mosier, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, John Wiley & Sons, Ltd, 2013, pp. 129–143.
184 D. D. Laskar, J. Zeng, L. Yan, S. Chen and B. Yang, *Ind. Crops Prod.*, 2013, **50**, 391–399.
185 H. L. Trajano, N. L. Engle, M. Foston, A. J. Ragauskas, T. J. Tschaplinski and C. E. Wyman, *Biotechnol. Biofuels*, 2013, **6**, 110.
186 X. Zhuang, Q. Yu, W. Wang, W. Qi, Q. Wang, X. Tan and Z. Yuan, *Appl. Biochem. Biotechnol.*, 2012, **168**, 206–218.
187 L. Yan, L. Zhang and B. Yang, *Biotechnol. Biofuels*, 2014, **7**, 76.
188 B. K. Avellar and W. G. Glasser, *Biomass Bioenergy*, 1998, **14**, 205–218.
189 W. G. Glasser and R. S. Wright, *Biomass Bioenergy*, 1998, **14**, 219–235.
190 M. Ibrahim and W. G. Glasser, *Bioresour. Technol.*, 1999, **70**, 181–192.
191 J. Li, G. Gellerstedt and K. Toven, *Bioresour. Technol.*, 2009, **100**, 2556–2561.
192 J. Li, G. Henriksson and G. Gellerstedt, *Bioresour. Technol.*, 2007, **98**, 3061–3068.
193 H. Heikkinen, T. Elder, H. Maaheimo, S. Rovio, J. Rahikainen, K. Kruus and T. Tamminen, *J. Agric. Food Chem.*, 2014, **62**, 10437–10444.
194 S. Ando, I. Arai, K. Kiyoto and S. Hanai, *J. Ferment. Technol.*, 1986, **64**, 567–570.
195 K. M. Torr, D. J. van de Pas, E. Cazeils and I. D. Suckling, *Bioresour. Technol.*, 2011, **102**, 7608–7611.
196 P. Sannigrahi and A. J. Ragauskas, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, John Wiley & Sons, Ltd, 2013, pp. 201–222.
197 X. Zhao, K. Cheng and D. Liu, *Appl. Microbiol. Biotechnol.*, 2009, **82**, 815–827.
198 M.-F. Li, S. Yang and R.-C. Sun, *Bioresour. Technol.*, 2016, **200**, 971–980.
199 W. J. J. Huijgen, A. T. Smit, J. H. Reith and H. den Uil, *J. Chem. Technol. Biotechnol.*, 2011, **86**, 1428–1438.
200 J. Wildschut, A. T. Smit, J. H. Reith and W. J. J. Huijgen, *Bioresour. Technol.*, 2013, **135**, 58–66.
201 W. J. J. Huijgen, G. Telysheva, A. Arshanitsa, R. J. A. Gosselink and P. J. de Wild, *Ind. Crops Prod.*, 2014, **59**, 85–95.
202 F. Monteil-Rivera, M. Phuong, M. Ye, A. Halasz and J. Hawari, *Ind. Crops Prod.*, 2013, **41**, 356–364.
203 B. B. Hallac, Y. Pu and A. J. Ragauskas, *Energy Fuels*, 2010, **24**, 2723–2732.
204 C. S. Lancefield, I. Panovic, P. J. Deuss, K. Barta and N. J. Westwood, *Green Chem.*, 2017, **19**, 202–214.
205 P. J. Deuss, C. S. Lancefield, A. Narani, J. G. de Vries, N. J. Westwood and K. Barta, *Green Chem.*, 2017, **19**, 2774–2782.
206 C. Nitsos, R. Stoklosa, A. Karnaouri, D. Vörös, H. Lange, D. Hodge, C. Crestini, U. Rova and P. Christakopoulos, *ACS Sustainable Chem. Eng.*, 2016, **4**, 5181–5193.
207 M. G. Alriols, A. Tejado, M. Blanco, I. Mondragon and J. Labidi, *Chem. Eng. J.*, 2009, **148**, 106–114.
208 L. P. Novo, L. V. A. Gurgel, K. Marabezi and A. A. d. S. Curvelo, *Bioresour. Technol.*, 2011, **102**, 10040–10046.
209 F. Sun and H. Chen, *J. Chem. Technol. Biotechnol.*, 2007, **82**, 1039–1044.
210 L. R. Limited, The Glycell™ Process, http://www.leafresources.com.au/Technology-Overview/The-Glycell-Process/, accessed December 2016, 2016.
211 T. vom Stein, P. M. Grande, H. Kayser, F. Sibilla, W. Leitner and P. Dominguez de Maria, *Green Chem.*, 2011, **13**, 1772–1777.
212 P. M. Grande, J. Viell, N. Theyssen, W. Marquardt, P. Dominguez de Maria and W. Leitner, *Green Chem.*, 2015, **17**, 3533–3539.
213 J. Quesada-Medina, F. J. López-Cremades and P. Olivares-Carrillo, *Bioresour. Technol.*, 2010, **101**, 8252–8260.
214 J. Snelders, E. Dornez, B. Benjelloun-Mlayah, W. J. J. Huijgen, P. J. de Wild, R. J. A. Gosselink, J. Gerritsma and C. M. Courtin, *Bioresour. Technol.*, 2014, **156**, 275–282.
215 F. Abdelkafi, H. Ammar, B. Rousseau, M. Tessier, R. El Gharbi and A. Fradet, *Biomacromolecules*, 2011, **12**, 3895–3902.
216 M. Delmas, *Chem. Eng. Technol.*, 2008, **31**, 792–797.
217 R. Katahira, A. Mittal, K. McKinney, P. N. Ciesielski, B. S. Donohoe, S. K. Black, D. K. Johnson, M. J. Biddy and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2014, **2**, 1364–1376.
218 W. J. Huijgen, J. H. Reith and H. den Uil, *Ind. Eng. Chem. Res.*, 2010, **49**, 10132–10140.
219 L. Jiménez, M. J. de la Torre, J. L. Bonilla and J. L. Ferrer, *Process Biochem.*, 1998, **33**, 401–408.
220 J. J. Bozell, S. K. Black, M. Myers, D. Cahill, W. P. Miller and S. Park, *Biomass Bioenergy*, 2011, **35**, 4197–4208.
221 T. J. Mcdonough, *Solvent Pulping Symposium*, 1992, **76**, 1–17.
222 E. K. Pye and J. Lora, *Tappi J.*, 1991, **74**, 113–118.
223 S. Bauer, H. Sorek, V. D. Mitchell, A. B. Ibáñez and D. E. Wemmer, *J. Agric. Food Chem.*, 2012, **60**, 8203–8212.
224 D. V. Evtuguin, C. P. Neto, A. M. S. Silva, P. M. Domingues, F. M. L. Amado, D. Robert and O. Faix, *J. Agric. Food Chem.*, 2001, **49**, 4252–4261.
225 G.-H. Delmas, B. Benjelloun-Mlayah, Y. L. Bigot and M. Delmas, *J. Appl. Polym. Sci.*, 2011, **121**, 491–501.
226 L. Shuai, M. T. Amiri, Y. M. Questell-Santiago, F. Héroguel, Y. Li, H. Kim, R. Meilan, C. Chapple, J. Ralph and J. S. Luterbacher, *Science*, 2016, **354**, 329–333.
227 G. H. van der Klashorst and S. A. Jackson, *J. Wood Chem. Technol.*, 1989, **9**, 1–15.
228 G. H. Van der Klashorst, *J. Wood Chem. Technol.*, 1988, **8**, 209–220.
229 G. H. van der Klashorst, in *Lignin – Properties and Materials*, American Chemical Society, 1989, ch. 26, vol. 397, pp. 346–360.

230 L. Shuai and B. Saha, *Green Chem.*, 2017, **19**, 3752–3758.
231 S. Van den Bosch, W. Schutyser, R. Vanholme, T. Driessen, S. F. Koelewijn, T. Renders, B. De Meester, W. J. J. Huijgen, W. Dehaen, C. M. Courtin, B. Lagrain, W. Boerjan and B. F. Sels, *Energy Environ. Sci.*, 2015, **8**, 1748–1763.
232 M. V. Galkin and J. S. M. Samec, *ChemSusChem*, 2014, **7**, 2154–2158.
233 T. Parsell, S. Yohe, J. Degenstein, T. Jarrell, I. Klein, E. Gencer, B. Hewetson, M. Hurt, J. I. Kim, H. Choudhari, B. Saha, R. Meilan, N. Mosier, F. Ribeiro, W. N. Delgass, C. Chapple, H. I. Kenttamaa, R. Agrawal and M. M. Abu-Omar, *Green Chem.*, 2015, **17**, 1492–1499.
234 P. Ferrini and R. Rinaldi, *Angew. Chem., Int. Ed.*, 2014, **53**, 8634–8639.
235 Q. Song, F. Wang, J. Cai, Y. Wang, J. Zhang, W. Yu and J. Xu, *Energy Environ. Sci.*, 2013, **6**, 994–1007.
236 W. Schutyser, S. Van den Bosch, T. Renders, T. De Boe, S. F. Koelewijn, A. Dewaele, T. Ennaert, O. Verkinderen, B. Goderis, C. M. Courtin and B. F. Sels, *Green Chem.*, 2015, **17**, 5035–5045.
237 T. Renders, S. Van den Bosch, T. Vangeel, T. Ennaert, S.-F. Koelewijn, G. Van den Bossche, C. M. Courtin, W. Schutyser and B. F. Sels, *ACS Sustainable Chem. Eng.*, 2016, **4**, 6894–6904.
238 T. Renders, W. Schutyser, S. Van den Bosch, S.-F. Koelewijn, T. Vangeel, C. M. Courtin and B. F. Sels, *ACS Catal.*, 2016, 2055–2066.
239 E. M. Anderson, R. Katahira, M. Reed, M. G. Resch, E. M. Karp, G. T. Beckham and Y. Roman-Leshkov, *ACS Sustainable Chem. Eng.*, 2016, **4**, 6940–6950.
240 X. Huang, J. Zhu, T. I. Korányi, M. D. Boot and E. J. M. Hensen, *ChemSusChem*, 2016, **9**, 3262–3267.
241 X. Huang, O. M. Morales Gonzalez, J. Zhu, T. I. Koranyi, M. D. Boot and E. J. M. Hensen, *Green Chem.*, 2017, **19**, 175–187.
242 X. Huang, X. Ouyang, B. Hendriks, O. Morales, J. Zhu, T. I. Koranyi, M. Boot and E. J. M. Hensen, *Faraday Discuss.*, 2017, **202**, 141–156.
243 S. Van den Bosch, W. Schutyser, S. F. Koelewijn, T. Renders, C. M. Courtin and B. F. Sels, *Chem. Commun.*, 2015, **51**, 13158–13161.
244 N. Yan, C. Zhao, P. J. Dyson, C. Wang, L. T. Liu and Y. Kou, *ChemSusChem*, 2008, **1**, 626–629.
245 C. Chesi, I. B. D. de Castro, M. T. Clough, P. Ferrini and R. Rinaldi, *ChemCatChem*, 2016, **8**, 2079–2088.
246 A. Brandt, J. Grasvik, J. P. Hallett and T. Welton, *Green Chem.*, 2013, **15**, 550–583.
247 C. Li, B. Knierim, C. Manisseri, R. Arora, H. V. Scheller, M. Auer, K. P. Vogel, B. A. Simmons and S. Singh, *Bioresour. Technol.*, 2010, **101**, 4900–4906.
248 T. Balensiefer, H. Schroeder, S. Freyer, G. D'Andola and K. Massonne, US 2010/0081798 A1, 2010.
249 T. C. R. Brennan, S. Datta, H. W. Blanch, B. A. Simmons and B. M. Holmes, *BioEnergy Res.*, 2010, **3**, 123–133.
250 K. C. Badgujar and B. M. Bhanage, *Bioresour. Technol.*, 2015, **178**, 2–18.
251 N. Sathitsuksanoh, K. M. Holtman, D. J. Yelle, T. Morgan, V. Stavila, J. Pelton, H. Blanch, B. A. Simmons and A. George, *Green Chem.*, 2014, **16**, 1236–1247.
252 A. Brandt, L. Chen, B. E. van Dongen, T. Welton and J. P. Hallett, *Green Chem.*, 2015, **17**, 5019–5034.
253 A. Brandt, F. Gschwend, P. Fennell, T. Lammens, B. Tan, J. Weale and J. Hallett, *Green Chem.*, 2017, **19**, 3078–3102.
254 P. Verdia, A. Brandt, J. P. Hallett, M. J. Ray and T. Welton, *Green Chem.*, 2014, **16**, 1617–1627.
255 S. H. Lee, T. V. Doherty, R. J. Linhardt and J. S. Dordick, *Biotechnol. Bioeng.*, 2009, **102**, 1368–1376.
256 L. Weigand, S. Mostame, A. Brandt, T. Welton and J. Hallett, *Faraday Discuss.*, 2017, **202**, 331–349.
257 J.-L. Wen, T.-Q. Yuan, S.-L. Sun, F. Xu and R.-C. Sun, *Green Chem.*, 2014, **16**, 181–190.
258 A. George, K. Tran, T. J. Morgan, P. I. Benke, C. Berrueco, E. Lorente, B. C. Wu, J. D. Keasling, B. A. Simmons and B. M. Holmes, *Green Chem.*, 2011, **13**, 3375–3385.
259 A. George, A. Brandt, K. Tran, S. M. S. N. S. Zahari, D. Klein-Marcuschamer, N. Sun, N. Sathitsuksanoh, J. Shi, V. Stavila, R. Parthasarathi, S. Singh, B. M. Holmes, T. Welton, B. A. Simmons and J. P. Hallett, *Green Chem.*, 2015, **17**, 1728–1734.
260 J. Rencoret, G. Marques, A. Gutiérrez, L. Nieto, J. Jiménez-Barbero, Á. T. Martínez and J. C. del Río, *Ind. Crops Prod.*, 2009, **30**, 137–143.
261 R. El Hage, N. Brosse, L. Chrusciel, C. Sanchez, P. Sannigrahi and A. Ragauskas, *Polym. Degrad. Stab.*, 2009, **94**, 1632–1638.
262 K. M. Holtman, H. m. Chang, H. Jameel and J. F. Kadla, *J. Wood Chem. Technol.*, 2006, **26**, 21–34.
263 K. M. Holtman, H.-m. Chang and J. F. Kadla, *J. Agric. Food Chem.*, 2004, **52**, 720–726.
264 V. S. Chang, M. Nagwani, C.-H. Kim and M. T. Holtzapple, *Appl. Biochem. Biotechnol.*, 2001, **94**, 1–28.
265 J. M. Gould, *Biotechnol. Bioeng.*, 1985, **27**, 225–231.
266 M. J. Selig, T. B. Vinzant, M. E. Himmel and S. R. Decker, *Appl. Biochem. Biotechnol.*, 2009, **155**, 94–103.
267 G. Banerjee, S. Car, T. Liu, D. L. Williams, S. L. Meza, J. D. Walton and D. B. Hodge, *Biotechnol. Bioeng.*, 2012, **109**, 922–931.
268 J. Luterbacher, D. M. Alonso and J. Dumesic, *Green Chem.*, 2014, **16**, 4816–4838.
269 R. A. Antonoplis, H. W. Blanch, R. P. Freitas, A. F. Sciamanna and C. R. Wilke, *Biotechnol. Bioeng.*, 1983, **25**, 2757–2773.
270 G. J. Ritter, R. L. Mitchell and R. M. Seborg, *J. Am. Chem. Soc.*, 1933, **55**, 2989–2991.
271 C. W. Dence, in *Methods in LigninChemistry*, ed. S. Lin and C. Dence, Springer Berlin Heidelberg, 1992, ch. 3, pp. 33–61.
272 R. D. Hatfield, H. J. G. Jung, J. Ralph, D. R. Buxton and P. J. Weimer, *J. Sci. Food Agric.*, 1994, **65**, 51–58.
273 Y. Lee, P. Iyer and R. W. Torget, in *Recent progress in bioconversion of lignocellulosics*, Springer, 1999, pp. 93–115.
274 P. Lenihan, A. Orozco, E. O'neill, M. Ahmad, D. Rooney and G. Walker, *Chem. Eng. J.*, 2010, **156**, 395–403.

275 P. Sannigrahi, D. H. Kim, S. Jung and A. Ragauskas, *Energy Environ. Sci.*, 2011, **4**, 1306–1310.
276 H. L. Trajano and C. E. Wyman, in *Aqueous Pretreatment of Plant Biomass for Biological and Chemical Conversion to Fuels and Chemicals*, ed. C. E. Wyman, John Wiley & Sons, Ltd, 2013, pp. 103–128.
277 I. van Zandvoort, Y. Wang, C. B. Rasrendra, E. R. van Eck, P. C. Bruijnincx, H. J. Heeres and B. M. Weckhuysen, *ChemSusChem*, 2013, **6**, 1745–1758.
278 I. van Zandvoort, E. J. Koers, M. Weingarth, P. C. Bruijnincx, M. Baldus and B. M. Weckhuysen, *Green Chem.*, 2015, **17**, 4383–4392.
279 J. S. Luterbacher, J. M. Rand, D. M. Alonso, J. Han, J. T. Youngquist, C. T. Maravelias, B. F. Pfleger and J. A. Dumesic, *Science*, 2014, **343**, 277–280.
280 D. M. Alonso, S. G. Wettstein, M. A. Mellmer, E. I. Gurbuz and J. A. Dumesic, *Energy Environ. Sci.*, 2013, **6**, 76–80.
281 J. S. Luterbacher, A. Azarpira, A. H. Motagamwala, F. C. Lu, J. Ralph and J. A. Dumesic, *Energy Environ. Sci.*, 2015, **8**, 2657–2663.
282 C. Sievers, M. B. Valenzuela-Olarte, T. Marzialetti, I. Musin, P. K. Agrawal and C. W. Jones, *Ind. Eng. Chem. Res.*, 2009, **48**, 1277–1286.
283 C. Li, Q. Wang and Z. K. Zhao, *Green Chem.*, 2008, **10**, 177–182.
284 B. Li, I. Filpponen and D. S. Argyropoulos, *Ind. Eng. Chem. Res.*, 2010, **49**, 3126–3136.
285 J. B. Binder and R. T. Raines, *Proc. Natl. Acad. Sci. U. S. A.*, 2010, **107**, 4516–4521.
286 N. Meine, R. Rinaldi and F. Schüth, *ChemSusChem*, 2012, **5**, 1449–1454.
287 M. Kaldstrom, N. Meine, C. Fares, R. Rinaldi and F. Schüth, *Green Chem.*, 2014, **16**, 2454–2462.
288 M. D. Kaufman Rechulski, M. Käldström, U. Richter, F. Schüth and R. Rinaldi, *Ind. Eng. Chem. Res.*, 2015, **54**, 4581–4592.
289 L. Schneider, Y. Dong, J. Haverinen, M. Jaakkola and U. Lassi, *Biomass Bioenergy*, 2016, **91**, 134–142.
290 M. Kaldstrom, N. Meine, C. Fares, F. Schüth and R. Rinaldi, *Green Chem.*, 2014, **16**, 3528–3538.
291 G. Calvaruso, M. T. Clough and R. Rinaldi, *Green Chem.*, 2017, **19**, 2803–2811.
292 G. Calvaruso, M. T. Clough, M. D. Kaufman Rechulski and R. Rinaldi, *ChemCatChem*, 2017, **9**, 2691–2700.
293 R. Katahira, A. Mittal, K. McKinney, X. Chen, M. P. Tucker, D. K. Johnson and G. T. Beckham, *ACS Sustainable Chem. Eng.*, 2016, **4**, 1474–1486.
294 P. Sannigrahi and A. J. Ragauskas, *J. Biobased Mater. Bioenergy*, 2011, **5**, 514–519.
295 J. I. Santos, R. Martín-Sampedro, Ú. Fillat, J. M. Oliva, M. J. Negro, M. Ballesteros, M. E. Eugenio and D. Ibarra, *Int. J. Polym. Sci.*, 2015, **2015**, 1–11.
296 P. Sannigrahi, Y. Pu and A. Ragauskas, *Curr. Opin. Environ. Sustain.*, 2010, **2**, 383–393.
297 R. El Hage, L. Chrusciel, L. Desharnais and N. Brosse, *Bioresour. Technol.*, 2010, **101**, 9321–9329.
298 R. Samuel, S. Cao, B. K. Das, F. Hu, Y. Pu and A. J. Ragauskas, *RSC Adv.*, 2013, **3**, 5305–5309.
299 R. Samuel, Y. Pu, B. Raman and A. J. Ragauskas, *Appl. Biochem. Biotechnol.*, 2010, **162**, 62–74.
300 S. Cao, Y. Pu, M. Studer, C. Wyman and A. J. Ragauskas, *RSC Adv.*, 2012, **2**, 10925–10936.
301 S. M. Shevchenko, R. P. Beatson and J. N. Saddler, *Appl. Biochem. Biotechnol.*, 1999, **79**, 867–876.
302 X. Chen, E. Kuhn, E. W. Jennings, R. Nelson, L. Tao, M. Zhang and M. P. Tucker, *Energy Environ. Sci.*, 2016, **9**, 1237–1245.
303 X. Chen, J. Shekiro, T. Pschorn, M. Sabourin, M. P. Tucker and L. Tao, *Biotechnol. Biofuels*, 2015, **8**, 1.
304 X. Chen, J. Shekiro, T. Pschorn, M. Sabourin, L. Tao, R. Elander, S. Park, E. Jennings, R. Nelson and O. Trass, *Biotechnol. Biofuels*, 2014, **7**, 1.
305 J. Vanneste, T. Ennaert, A. Vanhulsel and B. Sels, *ChemSusChem*, 2017, **10**, 14–31.
306 M. J. Bussemaker and D. Zhang, *Ind. Eng. Chem. Res.*, 2013, **52**, 3563–3580.
307 Q. Zhang, M. Benoit, K. De Oliveira Vigier, J. Barrault, G. Jegou, M. Philippe and F. Jerome, *Green Chem.*, 2013, **15**, 963–969.
308 T. Tana, Z. Zhang, L. Moghaddam, D. W. Rackemann, J. Rencoret, A. Gutierrez, J. C. del Río and W. O. Doherty, *ACS Sustainable Chem. Eng.*, 2016, **4**, 5483–5494.
309 K. H. Kim, B. A. Simmons and S. Singh, *Green Chem.*, 2017, **19**, 215–224.
310 J. Kong, M. He, J. A. Lercher and C. Zhao, *Chem. Commun.*, 2015, **51**, 17580–17583.
311 H.-m. Chang, B. Cowling Ellis and W. Brown, *Holzforschung*, 1975, **29**, 153–159.
312 Z. Hu, T.-F. Yeh, H.-m. Chang, Y. Matsumoto and F. Kadla John, *Holzforschung*, 2006, **60**, 389.
313 B. Jiang, T. Cao, F. Gu, W. Wu and Y. Jin, *ACS Sustainable Chem. Eng.*, 2017, **5**, 342–349.
314 T. Ikeda, K. Holtman, J. F. Kadla, H.-m. Chang and H. Jameel, *J. Agric. Food Chem.*, 2002, **50**, 129–135.
315 A. Guerra, I. Filpponen, L. A. Lucia, C. Saquing, S. Baumberger and D. S. Argyropoulos, *J. Agric. Food Chem.*, 2006, **54**, 5939–5947.
316 A. Guerra, I. Filpponen, L. A. Lucia and D. S. Argyropoulos, *J. Agric. Food Chem.*, 2006, **54**, 9696–9705.
317 A. Guerra, A. Lucia Lucian and S. Argyropoulos Dimitris, *Holzforschung*, 2008, **62**, 24.
318 A. Guerra, M. Norambuena, J. Freer and D. S. Argyropoulos, *J. Nat. Prod.*, 2008, **71**, 836–841.
319 A. V. Bridgwater, *Biomass Bioenergy*, 2012, **38**, 68–94.
320 C. Liu, H. Wang, A. M. Karim, J. Sun and Y. Wang, *Chem. Soc. Rev.*, 2014, **43**, 7594–7623.
321 D. A. Ruddy, J. A. Schaidle, J. R. Ferrell Iii, J. Wang, L. Moens and J. E. Hensley, *Green Chem.*, 2014, **16**, 454–490.
322 S. Xiu and A. Shahbazi, *Renewable Sustainable Energy Rev.*, 2012, **16**, 4406–4414.
323 M. Saidi, F. Samimi, D. Karimipourfard, T. Nimmanwudipong, B. C. Gates and M. R. Rahimpour, *Energy Environ. Sci.*, 2014, **7**, 103–129.

324 J. Jae, G. A. Tompsett, Y.-C. Lin, T. R. Carlson, J. Shen, T. Zhang, B. Yang, C. E. Wyman, W. C. Conner and G. W. Huber, *Energy Environ. Sci.*, 2010, **3**, 358–365.
325 A. G. Gayubo, B. Valle, A. T. Aguayo, M. Olazar and J. Bilbao, *J. Chem. Technol. Biotechnol.*, 2010, **85**, 132–144.
326 C. A. Mullen and A. A. Boateng, *Energy Fuels*, 2008, **22**, 2104–2109.
327 L. Ingram, D. Mohan, M. Bricka, P. Steele, D. Strobel, D. Crocker, B. Mitchell, J. Mohammad, K. Cantrell and C. U. Pittman, *Energy Fuels*, 2008, **22**, 614–625.
328 E. B. Hassan, H. Abou-Yousef, P. Steele and E. El-Giar, *Energy Sources, Part A*, 2016, **38**, 43–50.
329 B. Scholze, C. Hanser and D. Meier, *J. Anal. Appl. Pyrolysis*, 2001, **58**, 387–400.
330 B. Scholze and D. Meier, *J. Anal. Appl. Pyrolysis*, 2001, **60**, 41–54.
331 R. Bayerbach and D. Meier, *J. Anal. Appl. Pyrolysis*, 2009, **85**, 98–107.
332 R. Bayerbach, V. D. Nguyen, U. Schurr and D. Meier, *J. Anal. Appl. Pyrolysis*, 2006, **77**, 95–101.
333 T. Phongpreecha, N. C. Hool, R. J. Stoklosa, A. S. Klett, C. E. Foster, A. Bhalla, D. Holmes, M. C. Thies and D. B. Hodge, *Green Chem.*, 2017, **19**, 5131–5143.
334 J. Ralph and L. L. Landucci, in *Lignin and lignans: advances in chemistry*, ed. C. Heitner, D. R. Dimmel and J. A. Schmidt, CRC Press, 2011, pp. 137–244.
335 C. Rolando, B. Monties and C. Lapierre, in *Methods in Lignin Chemistry*, ed. S. Y. Lin and C. W. Dence, Springer Berlin Heidelberg, Berlin, Heidelberg, 1992, pp. 334–349.
336 C. Lapierre, B. Monties and C. Rolando, *Holzforschung*, 1986, **40**, 113.
337 R. Katahira and F. Nakatsubo, *J. Wood Sci.*, 2001, **47**, 378–382.
338 P. C. R. Pinto, C. E. Costa and A. E. Rodrigues, *Ind. Eng. Chem. Res.*, 2013, **52**, 4421–4428.
339 The average values were calculated based on the information provided in Table 1 in the study of Barta *et al.* L1–L18 correspond to the self-synthesized organosolv lignins, L19–L22 to the technical organosolv lignins.
340 M. H. Studer, J. D. DeMartini, M. F. Davis, R. W. Sykes, B. Davison, M. Keller, G. A. Tuskan and C. E. Wyman, *Proc. Natl. Acad. Sci. U. S. A.*, 2011, **108**, 6300–6305.
341 C. L. Cass, A. A. Lavell, N. Santoro, C. E. Foster, S. D. Karlen, R. A. Smith, J. Ralph, D. F. Garvin and J. C. Sedbrook, *Front. Recent Dev. Plant Sci.*, 2016, **7**, 708.
342 T. Renders, S. Van den Bosch, S. F. Koelewijn, W. Schutyser and B. F. Sels, *Energy Environ. Sci.*, 2017, **10**, 1551–1557.
343 To exemplify, we have calculated the liquid-to-solid (L/S) ratios used in batch- and FT-HWP of poplar in a study by Wyman and co-workers.[185] For batch-HWP, 0.44 g substrate was submerged in 7.88 mL of water, corresponding to an L/S-ratio of 18 mL g^{-1}. In the FT-HWP experiments, 1 g substrate was treated with a 20 mL min^{-1} water flow for either 12 or 192 min. This corresponds to L/S-ratios of respectively 240 and 3840 mL g^{-1}.
344 F. Chen and R. A. Dixon, *Nat. Biotechnol.*, 2007, **25**, 759–761.
345 C. G. Wilkerson, S. D. Mansfield, F. Lu, S. Withers, J. Y. Park, S. D. Karlen, E. Gonzales-Vigil, D. Padmakshan, F. Unda, J. Rencoret and J. Ralph, *Science*, 2014, **344**, 90.
346 K. H. Kim, T. Dutta, J. Ralph, S. D. Mansfield, B. A. Simmons and S. Singh, *Biotechnol. Biofuels*, 2017, **10**, 101.
347 N. D. Bonawitz and C. Chapple, *Curr. Opin. Biotechnol.*, 2013, **24**, 336–343.
348 N. D. Bonawitz, J. I. Kim, Y. Tobimatsu, P. N. Ciesielski, N. A. Anderson, E. Ximenes, J. Maeda, J. Ralph, B. S. Donohoe, M. Ladisch and C. Chapple, *Nature*, 2014, **509**, 376–380.
349 E. Feghali, G. Carrot, P. Thuery, C. Genre and T. Cantat, *Energy Environ. Sci.*, 2015, **8**, 2734–2743.
350 The total product yield, oil yield, and lignin conversion are usually defined as the fraction of lignin that is soluble (product yield) or not soluble (unconverted lignin) in the applied solvent after reaction, or in an extraction solvent such as ethyl acetate, dichloromethane or diethylether.
351 H. Luo, I. M. Klein, Y. Jiang, H. Zhu, B. Liu, H. I. Kenttämaa and M. M. Abu-Omar, *ACS Sustainable Chem. Eng.*, 2016, **4**, 2316–2322.
352 M. V. Galkin, A. T. Smit, E. Subbotina, K. A. Artemenko, J. Bergquist, W. J. J. Huijgen and J. S. M. Samec, *ChemSusChem*, 2016, **9**, 3280–3287.
353 C. Li, M. Zheng, A. Wang and T. Zhang, *Energy Environ. Sci.*, 2012, **5**, 6383–6390.
354 J. M. Pepper, W. F. Steck, R. Swoboda and J. C. Karapally, *Lignin Structure and Reactions*, American Chemical Society, 1966, ch. 16, vol. 59, pp. 238–248.
355 J. M. Pepper and P. Supathna, *Can. J. Chem.*, 1978, **56**, 899–902.
356 I. Klein, C. Marcum, H. Kenttamaa and M. M. Abu-Omar, *Green Chem.*, 2016, **18**, 2399–2405.
357 J. M. Pepper and Y. W. Lee, *Can. J. Chem.*, 1969, **47**, 723–727.
358 I. Klein, B. Saha and M. M. Abu-Omar, *Catal. Sci. Technol.*, 2015, **5**, 3242–3245.
359 J. M. Pepper and W. Steck, *Can. J. Chem.*, 1963, **41**, 2867–2875.
360 J. M. Pepper and Y. W. Lee, *Can. J. Chem.*, 1970, **48**, 477–479.
361 J. M. Pepper and R. W. Fleming, *Can. J. Chem.*, 1978, **56**, 896–898.
362 J. M. Pepper and M. D. Rahman, *Cellul. Chem. Technol.*, 1986, **21**, 233–239.
363 T. S. Kaplunova, K. A. Abduazimov, B. K. Pulatov, M. T. Ikramutdinova and M. F. Abidova, *Chem. Nat. Compd.*, 1993, **29**, 530–532.
364 J. Chen, F. Lu, X. Si, X. Nie, J. Chen, R. Lu and J. Xu, *ChemSusChem*, 2016, **9**, 3353–3360.
365 H. P. Godard, J. L. McCarthy and H. Hibbert, *J. Am. Chem. Soc.*, 1941, **63**, 3061–3066.
366 S. Van den Bosch, T. Renders, S. Kennis, S.-F. Koelewijn, G. Van den Bossche, T. Vangeel, A. Deneyer, D. Depuydt, C. M. Courtin, J. Thevelein, W. Schutyser and B. F. Sels, *Green Chem.*, 2017, **19**, 3313–3326.
367 F. Lu, S. D. Karlen, M. Regner, H. Kim, S. A. Ralph, R.-C. Sun, K.-i. Kuroda, M. A. Augustin, R. Mawson and H. Sabarez, *BioEnergy Res.*, 2015, **8**, 934–952.

368 Y. Liu, L. Chen, T. Wang, Q. Zhang, C. Wang, J. Yan and L. Ma, *ACS Sustainable Chem. Eng.*, 2015, **3**, 1745–1755.
369 T. D. Matson, K. Barta, A. V. Iretskii and P. C. Ford, *J. Am. Chem. Soc.*, 2011, **133**, 14090–14097.
370 Q. Xia, Z. Chen, Y. Shao, X. Gong, H. Wang, X. Liu, S. F. Parker, X. Han, S. Yang and Y. Wang, *Nat. Commun.*, 2016, **7**, 11162.
371 V. E. Tarabanko, K. L. Kaygorodov, E. A. Skiba, N. Tarabanko, Y. V. Chelbina, O. V. Baybakova, B. N. Kuznetsov and L. Djakovitch, *J. Wood Chem. Technol.*, 2017, **37**, 43–51.
372 J. M. Pepper, B. W. Casselman and J. C. Karapally, *Can. J. Chem.*, 1967, **45**, 3009–3012.
373 E. Minami and S. Saka, *J. Wood Sci.*, 2003, **49**, 0073–0078.
374 E. Minami and S. Saka, *J. Wood Sci.*, 2005, **51**, 395–400.
375 P. J. Deuss, C. W. Lahive, C. S. Lancefield, N. J. Westwood, P. C. J. Kamer, K. Barta and J. G. de Vries, *ChemSusChem*, 2016, **9**, 2974–2981.
376 C. W. Lahive, P. J. Deuss, C. S. Lancefield, Z. H. Sun, D. B. Cordes, C. M. Young, F. Tran, A. M. Z. Slawin, J. G. de Vries, P. C. J. Kamer, N. J. Westwood and K. Barta, *J. Am. Chem. Soc.*, 2016, **138**, 8900–8911.
377 R. Jastrzebski, S. Constant, C. S. Lancefield, N. J. Westwood, B. M. Weckhuysen and P. C. A. Bruijnincx, *ChemSusChem*, 2016, **9**, 2074–2079.
378 A. Kaiho, M. Kogo, R. Sakai, K. Saito and T. Watanabe, *Green Chem.*, 2015, **17**, 2780–2783.
379 In reaction with eucalyptus, a monomer yield of 3 wt% was obtained on wood basis, which corresponds to a yield of 10 to 13 wt% on lignin basis if a lignin content of 24 to 29 wt% (see *Ind. Eng. Chem. Res.*, 2008, **47**, 8542–8549) is suggested.
380 C. S. Lancefield, O. S. Ojo, F. Tran and N. J. Westwood, *Angew. Chem., Int. Ed.*, 2015, **54**, 258–262.
381 O. Faix and D. Meier, *Holz Roh- Werkst.*, 1989, **47**, 67–72.
382 A. McVeigh, F. P. Bouxin, M. C. Jarvis and S. D. Jackson, *Catal. Sci. Technol.*, 2016, **6**, 4142–4150.
383 J.-Y. Kim, J. Park, U.-J. Kim and J. W. Choi, *Energy Fuels*, 2015, **29**, 5154–5163.
384 Y. Ye, Y. Zhang, J. Fan and J. Chang, *Bioresour. Technol.*, 2012, **118**, 648–651.
385 Q. Song, F. Wang and J. Xu, *Chem. Commun.*, 2012, **48**, 7019–7021.
386 R. Shu, J. Long, Z. Yuan, Q. Zhang, T. Wang, C. Wang and L. Ma, *Bioresour. Technol.*, 2015, **179**, 84–90.
387 J. Long, Y. Xu, T. Wang, Z. Yuan, R. Shu, Q. Zhang and L. Ma, *Appl. Energy*, 2015, **141**, 70–79.
388 H. Konnerth, J. Zhang, D. Ma, M. H. G. Prechtl and N. Yan, *Chem. Eng. Sci.*, 2015, **123**, 155–163.
389 K. Barta, G. R. Warner, E. S. Beach and P. T. Anastas, *Green Chem.*, 2014, **16**, 191–196.
390 C. J. Coscia, W. J. Schubert and F. F. Nord, *J. Org. Chem.*, 1961, **26**, 5085–5091.
391 A. Olcay, *J. Org. Chem.*, 1962, **27**, 1783–1786.
392 T. P. Schultz, C.-L. Chen and I. S. Goldstein, *J. Wood Chem. Technol.*, 1982, **2**, 33–46.
393 L.-T. Liu, B. Zhang, J. Li, D. Ma and Y. Kou, *Acta Phys.-Chim. Sin.*, 2012, **28**, 2343–2348.
394 D. Forchheim, U. Hornung, P. Kempe, A. Kruse and D. Steinbach, *Int. J. Chem. Eng.*, 2012, **2012**, 1–8.
395 X. Ma, Y. Tian, W. Hao, R. Ma and Y. Li, *Appl. Catal., A*, 2014, **481**, 64–70.
396 J. Zhang, J. Teo, X. Chen, H. Asakura, T. Tanaka, K. Teramura and N. Yan, *ACS Catal.*, 2014, **4**, 1574–1583.
397 J. Zhang, H. Asakura, J. van Rijn, J. Yang, P. Duchesne, B. Zhang, X. Chen, P. Zhang, M. Saeys and N. Yan, *Green Chem.*, 2014, **16**, 2432–2437.
398 J.-Y. Kim, J. Park, H. Hwang, J. K. Kim, I. K. Song and J. W. Choi, *J. Anal. Appl. Pyrolysis*, 2015, **113**, 99–106.
399 V. Molinari, G. Clavel, M. Graglia, M. Antonietti and D. Esposito, *ACS Catal.*, 2016, **6**, 1663–1670.
400 R. Shu, J. Long, Y. Xu, L. Ma, Q. Zhang, T. Wang, C. Wang, Z. Yuan and Q. Wu, *Bioresour. Technol.*, 2016, **200**, 14–22.
401 J. Zakzeski, A. L. Jongerius, P. C. A. Bruijnincx and B. M. Weckhuysen, *ChemSusChem*, 2012, **5**, 1602–1609.
402 X. Wang and R. Rinaldi, *ChemSusChem*, 2012, **5**, 1455–1466.
403 A. Kloekhorst and H. J. Heeres, *ACS Sustainable Chem. Eng.*, 2015, **3**, 1905–1914.
404 C. R. Kumar, N. Anand, A. Kloekhorst, C. Cannilla, G. Bonura, F. Frusteri, K. Barta and H. J. Heeres, *Green Chem.*, 2015, **17**, 4921–4930.
405 A. Narani, R. K. Chowdari, C. Cannilla, G. Bonura, F. Frusteri, H. J. Heeres and K. Barta, *Green Chem.*, 2015, **17**, 5046–5057.
406 A. Kloekhorst and H. J. Heeres, *Catal. Sci. Technol.*, 2016, **6**, 7053–7067.
407 D. Meier, R. Ante and O. Faix, *Bioresour. Technol.*, 1992, **40**, 171–177.
408 D. Meier, J. Berns, C. Grünwald and O. Faix, *J. Anal. Appl. Pyrolysis*, 1993, **25**, 335–347.
409 A. Oasmaa, R. Alén and D. Meier, *Bioresour. Technol.*, 1993, **45**, 189–194.
410 M. A. Ratcliff, D. K. Johnson, F. L. Posey and H. L. Chum, *Appl. Biochem. Biotechnol.*, 1988, **17**, 151–160.
411 S. Kasakov, H. Shi, D. M. Camaioni, C. Zhao, E. Barath, A. Jentys and J. A. Lercher, *Green Chem.*, 2015, **17**, 5079–5090.
412 D. D. Laskar, M. P. Tucker, X. Chen, G. L. Helms and B. Yang, *Green Chem.*, 2014, **16**, 897–910.
413 H. Wang, H. Ruan, H. Pei, H. Wang, X. Chen, M. P. Tucker, J. R. Cort and B. Yang, *Green Chem.*, 2015, **17**, 5131–5135.
414 H. Wang, H. Ruan, M. Feng, Y. Qin, H. Job, L. Luo, C. Wang, M. H. Engelhard, E. Kuhn, X. Chen, M. P. Tucker and B. Yang, *ChemSusChem*, 2017, **10**, 1846–1856.
415 X. Wang and R. Rinaldi, *Catal. Today*, 2016, **269**, 48–55.
416 X. Huang, T. I. Korányi, M. D. Boot and E. J. M. Hensen, *ChemSusChem*, 2014, **7**, 2276–2288.
417 X. Huang, T. I. Koranyi, M. D. Boot and E. J. M. Hensen, *Green Chem.*, 2015, **17**, 4941–4950.
418 R. Ma, W. Hao, X. Ma, Y. Tian and Y. Li, *Angew. Chem., Int. Ed.*, 2014, **53**, 7310–7315.
419 X. Ma, R. Ma, W. Hao, M. Chen, F. Yan, K. Cui, Y. Tian and Y. Li, *ACS Catal.*, 2015, **5**, 4803–4813.
420 X. Ma, K. Cui, W. Hao, R. Ma, Y. Tian and Y. Li, *Bioresour. Technol.*, 2015, **192**, 17–22.

421 Y. Jiang, Z. Li, X. Tang, Y. Sun, X. Zeng, S. Liu and L. Lin, *Energy Fuels*, 2015, **29**, 1662–1668.
422 A. Kloekhorst, Y. Shen, Y. Yie, M. Fang and H. J. Heeres, *Biomass Bioenergy*, 2015, **80**, 147–161.
423 A. L. Jongerius, P. C. A. Bruijnincx and B. M. Weckhuysen, *Green Chem.*, 2013, **15**, 3049–3056.
424 L. Liguori and T. Barth, *J. Anal. Appl. Pyrolysis*, 2011, **92**, 477–484.
425 W. Xu, S. J. Miller, P. K. Agrawal and C. W. Jones, *ChemSusChem*, 2012, **5**, 667–675.
426 X. Huang, C. Atay, T. I. Korányi, M. D. Boot and E. J. M. Hensen, *ACS Catal.*, 2015, **5**, 7359–7370.
427 L. Chen, T. I. Koranyi and E. J. M. Hensen, *Chem. Commun.*, 2016, **52**, 9375–9378.
428 A. Toledano, L. Serrano, J. Labidi, A. Pineda, A. M. Balu and R. Luque, *ChemCatChem*, 2013, **5**, 977–985.
429 A. Toledano, L. Serrano, A. M. Balu, R. Luque, A. Pineda and J. Labidi, *ChemSusChem*, 2013, **6**, 529–536.
430 A. Toledano, L. Serrano, A. Pineda, A. A. Romero, R. Luque and J. Labidi, *Appl. Catal., B*, 2014, **145**, 43–55.
431 G. Zhu, X. Qiu, Y. Zhao, Y. Qian, Y. Pang and X. Ouyang, *Bioresour. Technol.*, 2016, **218**, 718–722.
432 J. A. Onwudili and P. T. Williams, *Green Chem.*, 2014, **16**, 4740–4748.
433 S. K. Singh and J. D. Ekhe, *Catal. Sci. Technol.*, 2015, **5**, 2117–2124.
434 E. Feghali and T. Cantat, US 2017/0137446 A1, 2017.
435 C. Lapierre, *Lignin and Lignans*, CRC Press, 2010, pp. 11–48.
436 H. Lange, S. Decina and C. Crestini, *Eur. Polym. J.*, 2013, **49**, 1151–1173.
437 S. K. Hanson and R. T. Baker, *Acc. Chem. Res.*, 2015, **48**, 2037–2048.
438 G. Chatel and R. D. Rogers, *ACS Sustainable Chem. Eng.*, 2014, **2**, 322–339.
439 R. E. Key and J. J. Bozell, *ACS Sustainable Chem. Eng.*, 2016, **4**, 5123–5135.
440 J. J. Bozell, in *Selective Catalysis for Renewable Feedstocks andChemicals*, ed. K. M. Nicholas, Springer International Publishing, Cham, 2014, pp. 229–255.
441 R. Behling, S. Valange and G. Chatel, *Green Chem.*, 2016, **18**, 1839–1854.
442 L. Das, P. Kolar and R. Sharma-Shivappa, *Biofuels*, 2012, **3**, 155–166.
443 M. Fache, B. Boutevin and S. Caillol, *ACS Sustainable Chem. Eng.*, 2016, **4**, 35–46.
444 P. C. Rodrigues Pinto, E. A. Borges da Silva and A. E. Rodrigues, in *Biomass Conversion: The Interface of Biotechnology, Chemistry and Materials Science*, ed. C. Baskar, S. Baskar and R. S. Dhillon, Springer Berlin Heidelberg, Berlin, Heidelberg, 2012, pp. 381–420.
445 A. L. Mathias and A. E. Rodrigues, *Holzforschung*, 1995, **49**, 273–278.
446 C. Fargues, Á. Mathias and A. Rodrigues, *Ind. Eng. Chem. Res.*, 1996, **35**, 28–36.
447 P. Sridhar, J. D. Araujo and A. E. Rodrigues, *Catal. Today*, 2005, **105**, 574–581.
448 J. D. P. Araújo, C. A. Grande and A. E. Rodrigues, *Chem. Eng. Res. Des.*, 2010, **88**, 1024–1032.
449 V. E. Tarabanko, N. A. Fomova, B. N. Kuznetsov, N. M. Ivanchenko and A. V. Kudryashev, *React. Kinet. Catal. Lett.*, 1995, **55**, 161–170.
450 S. Kagawa and M. Rokugawa, *Jpn. TAPPI J.*, 1971, **25**, 506–511.
451 J. C. Villar, A. Caperos and F. García-Ochoa, *Wood Sci. Technol.*, 2001, **35**, 245–255.
452 S. G. Santos, A. P. Marques, D. L. D. Lima, D. V. Evtuguin and V. I. Esteves, *Ind. Eng. Chem. Res.*, 2011, **50**, 291–298.
453 V. E. Taraban'ko, N. V. Koropatchinskaya, A. V. Kudryashev and B. N. Kuznetsov, *Russ. Chem. Bull.*, 1995, **44**, 367–371.
454 G. Wu, M. Heitz and E. Chornet, *Ind. Eng. Chem. Res.*, 1994, **33**, 718–723.
455 G. Wu and M. Heitz, *J. Wood Chem. Technol.*, 1995, **15**, 189–202.
456 F. G. Sales, C. A. M. Abreu and J. A. F. R. Pereira, *Braz. J. Chem. Eng.*, 2004, **21**, 211–218.
457 F. G. Sales, L. C. A. Maranhão, N. M. Lima Filho and C. A. M. Abreu, *Ind. Eng. Chem. Res.*, 2006, **45**, 6627–6631.
458 F. G. Sales, L. C. A. Maranhão, N. M. L. Filho and C. A. M. Abreu, *Chem. Eng. Sci.*, 2007, **62**, 5386–5391.
459 J. Zhang, H. Deng and L. Lin, *Molecules*, 2009, **14**, 2747.
460 H. Deng, L. Lin, Y. Sun, C. Pang, J. Zhuang, P. Ouyang, J. Li and S. Liu, *Energy Fuels*, 2009, **23**, 19–24.
461 H. Deng, L. Lin and S. Liu, *Energy Fuels*, 2010, **24**, 4797–4802.
462 H. Deng, L. Lin, Y. Sun, C. Pang, J. Zhuang, P. Ouyang, Z. Li and S. Liu, *Catal. Lett.*, 2008, **126**, 106.
463 B. Wang and Z. Long, *Appl. Mech. Mater.*, 2011, **80-81**, 350–354.
464 P. Gao, C. Li, H. Wang, X. Wang and A. Wang, *Chin. J. Catal.*, 2013, **34**, 1811–1815.
465 A. L. Mathias, M. I. Lopretti and A. E. Rodrigues, *J. Chem. Technol. Biotechnol.*, 1995, **64**, 225–234.
466 J. D. P. Araújo, C. A. Grande and A. E. Rodrigues, *Catal. Today*, 2009, **147**(supplement), S330–S335.
467 P. C. R. Pinto, E. A. B. da Silva and A. E. Rodrigues, *Ind. Eng. Chem. Res.*, 2010, **49**, 12311–12318.
468 P. C. Rodrigues Pinto, E. A. Borges da Silva and A. E. Rodrigues, *Ind. Eng. Chem. Res.*, 2011, **50**, 741–748.
469 H.-R. Bjørsvik and F. Minisci, *Org. Process Res. Dev.*, 1999, **3**, 330–340.
470 K. R. Alunga, Y. Y. Ye, S. R. Li, D. Wang and Y. Q. Liu, *Catal. Sci. Technol.*, 2015, **5**, 3746–3753.
471 A. Azarpira, J. Ralph and F. C. Lu, *BioEnergy Res.*, 2014, **7**, 78–86.
472 A. W. Pacek, P. Ding, M. Garrett, G. Sheldrake and A. W. Nienow, *Ind. Eng. Chem. Res.*, 2013, **52**, 8361–8372.
473 X. P. Ouyang, T. Ruan and X. Q. Qiu, *Fuel Process. Technol.*, 2016, **144**, 181–185.
474 T. Voitl and P. R. von Rohr, *ChemSusChem*, 2008, **1**, 763–769.
475 T. Voitl, M. V. Nagel and P. R. von Rohr, *Holzforschung*, 2010, **64**, 13–19.
476 T. Voitl and P. R. von Rohr, *Ind. Eng. Chem. Res.*, 2010, **49**, 520–525.

477 H. Werhan, J. M. Mir, T. Voitl and P. R. von Rohr, *Holzforschung*, 2011, **65**, 703–709.
478 H. Werhan, N. Assmann and P. R. von Rohr, *Chem. Eng. Process.*, 2013, **73**, 29–37.
479 W. Partenheimer, *Adv. Synth. Catal.*, 2009, **351**, 456–466.
480 A. R. Goncalves and U. Schuchardt, *Appl. Biochem. Biotechnol.*, 1999, **77-9**, 127–132.
481 R. S. Ma, M. Guo, K. T. Lin, V. R. Hebert, J. W. Zhang, M. P. Wolcott, M. Quintero, K. K. Ramasamy, X. W. Chen and X. Zhang, *Chem. – Eur. J.*, 2016, **22**, 10884–10891.
482 W. P. Deng, H. X. Zhang, X. J. Wu, R. S. Li, Q. H. Zhang and Y. Wang, *Green Chem.*, 2015, **17**, 5009–5018.
483 R. Prado, A. Brandt, X. Erdocia, J. Hallet, T. Welton and J. Labidi, *Green Chem.*, 2016, **18**, 834–841.
484 G. F. De Gregorio, R. Prado, C. Vriamont, X. Erdocia, J. Labidi, J. P. Hallett and T. Welton, *ACS Sustainable Chem. Eng.*, 2016, **4**, 6031–6036.
485 R. Prado, X. Erdocia, G. F. De Gregorio, J. Labidi and T. Welton, *ACS Sustainable Chem. Eng.*, 2016, **4**, 5277–5288.
486 K. Stark, N. Taccardi, A. Bosmann and P. Wasserscheid, *ChemSusChem*, 2010, **3**, 719–723.
487 S. W. Liu, Z. L. Shi, L. Li, S. T. Yu, C. X. Xie and Z. Q. Song, *RSC Adv.*, 2013, **3**, 5789–5793.
488 R. S. Ma, M. Guo and X. Zhang, *ChemSusChem*, 2014, **7**, 412–415.
489 I. Hasegawa, Y. Inoue, Y. Muranaka, T. Yasukawa and K. Mae, *Energy Fuels*, 2011, **25**, 791–796.
490 A. G. Demesa, A. Laari, I. Turunen and M. Sillanpaa, *Chem. Eng. Technol.*, 2015, **38**, 2270–2278.
491 S. Lotfi, D. C. Boffito and G. S. Patience, *ChemSusChem*, 2015, **8**, 3424–3432.
492 V. M. Roberts, V. Stein, T. Reiner, A. Lemonidou, X. B. Li and J. A. Lercher, *Chem. – Eur. J.*, 2011, **17**, 5939–5948.
493 J. M. Lavoie, W. Bare and M. Bilodeau, *Bioresour. Technol.*, 2011, **102**, 4917–4920.
494 R. Beauchet, F. Monteil-Rivera and J. M. Lavoie, *Bioresour. Technol.*, 2012, **121**, 328–334.
495 A. Toledano, L. Serrano and J. Labidi, *J. Chem. Technol. Biotechnol.*, 2012, **87**, 1593–1599.
496 L. Du, Z. Wang, S. G. Li, W. L. Song and W. G. Lin, *Int. J. Chem. React. Eng.*, 2013, **11**, 135–145.
497 R. W. Thring, *Biomass Bioenergy*, 1994, **7**, 125–130.
498 X. Erdocia, R. Prado, M. A. Corcuera and J. Labidi, *Biomass Bioenergy*, 2014, **66**, 379–386.
499 S. Kudo, Y. Hachiyama, Y. Takashima, J. Tahara, S. Idesh, K. Norinaga and J. Hayashi, *Energy Fuels*, 2014, **28**, 76–85.
500 U. Schuchardt, J. A. R. Rodrigues, A. Cotrim and J. L. M. Costa, *Bioresour. Technol.*, 1993, **44**, 123–129.
501 J. S. Kruger, N. S. Cleveland, S. T. Zhang, R. Katahira, B. A. Black, G. M. Chupka, T. Lammens, P. G. Hamilton, M. J. Biddy and G. T. Beckham, *ACS Catal.*, 2016, **6**, 1316–1328.
502 R. Chaudhary and P. L. Dhepe, *Green Chem.*, 2017, **19**, 778–788.
503 J. X. Long, Q. Zhang, T. J. Wang, X. H. Zhang, Y. Xu and L. L. Ma, *Bioresour. Technol.*, 2014, **154**, 10–17.
504 M. R. Sturgeon, M. H. O'Brien, P. N. Ciesielski, R. Katahira, J. S. Kruger, S. C. Chmely, J. Hamlin, K. Lawrence, G. B. Hunsinger, T. D. Foust, R. M. Baldwin, M. J. Biddy and G. T. Beckham, *Green Chem.*, 2014, **16**, 824–835.
505 M. M. Hepditch and R. W. Thring, *Can. J. Chem. Eng.*, 2000, **78**, 226–231.
506 X. H. Zhang, Q. Zhang, J. X. Long, Y. Xu, T. J. Wang, L. L. Ma and Y. P. Li, *BioResources*, 2014, **9**, 3347–3360.
507 B. Guvenatam, E. H. J. Heeres, E. A. Pidko and E. J. M. Hensen, *Catal. Today*, 2016, **269**, 9–20.
508 B. Guvenatam, E. H. J. Heeres, E. A. Pidko and E. J. M. Hensen, *Catal. Today*, 2016, **259**, 460–466.
509 A. K. Deepa and P. L. Dhepe, *RSC Adv.*, 2014, **4**, 12625–12629.
510 M. Kleinert and T. Barth, *Chem. Eng. Technol.*, 2008, **31**, 736–745.
511 M. Kleinert and T. Barth, *Energy Fuels*, 2008, **22**, 1371–1379.
512 J. R. Gasson, D. Forchheim, T. Sutter, U. Hornung, A. Kruse and T. Barth, *Ind. Eng. Chem. Res.*, 2012, **51**, 10595–10606.
513 R. J. A. Gosselink, W. Teunissen, J. E. G. van Dam, E. de Jong, G. Gellerstedt, E. L. Scott and J. P. M. Sanders, *Bioresour. Technol.*, 2012, **106**, 173–177.
514 A. K. Deepa and P. L. Dhepe, *ACS Catal.*, 2015, **5**, 365–379.
515 T. Yoshikawa, T. Yagi, S. Shinohara, T. Fukunaga, Y. Nakasaka, T. Tago and T. Masuda, *Fuel Process. Technol.*, 2013, **108**, 69–75.
516 T. Yoshikawa, S. Shinohara, T. Yagi, N. Ryumon, Y. Nakasaka, T. Tago and T. Masuda, *Appl. Catal., B*, 2014, **146**, 289–297.
517 Y. Y. Ye, J. Fan and J. Chang, *J. Anal. Appl. Pyrolysis*, 2012, **94**, 190–195.
518 Y. Y. Ye, Y. Zhang, J. Fan and J. Chang, *Ind. Eng. Chem. Res.*, 2012, **51**, 103–110.
519 H. S. Lee, J. Jae, J. M. Ha and D. J. Suh, *Bioresour. Technol.*, 2016, **203**, 142–149.
520 T. L. K. Yong and Y. Matsumura, *Ind. Eng. Chem. Res.*, 2013, **52**, 5626–5639.
521 W. Diono, M. Sasaki and M. Goto, *Chem. Eng. Process.*, 2008, **47**, 1609–1619.
522 H. Pinkowska, P. Wolak and A. Zlocinska, *Chem. Eng. J.*, 2012, **187**, 410–414.
523 B. Zhang, H. J. Huang and S. Ramaswamy, *Appl. Biochem. Biotechnol.*, 2008, **147**, 119–131.
524 Z. C. Jiang, T. He, J. M. Li and C. W. Hu, *Green Chem.*, 2014, **16**, 4257–4265.
525 L. B. Hu, Y. P. Luo, B. Cai, J. M. Li, D. M. Tong and C. W. Hu, *Green Chem.*, 2014, **16**, 3107–3116.
526 Y. Y. Lee and B. H. Lee, *J. Ind. Eng. Chem.*, 1998, **4**, 334–339.
527 X. Erdocia, R. Prado, J. Fernandez-Rodriguez and J. Labidi, *ACS Sustainable Chem. Eng.*, 2016, **4**, 1373–1380.
528 K. H. Kim, R. C. Brown, M. Kieffer and X. L. Bai, *Energy Fuels*, 2014, **28**, 6429–6437.
529 E. Dorrestijn, M. Kranenburg, D. Poinsot and P. Mulder, *Holzforschung*, 1999, **53**, 611–616.
530 W. Diono, M. Sasaki and M. Goto, *J. Mater. Cycles Waste Manage.*, 2011, **13**, 68–79.
531 D. J. Nowakowski, A. V. Bridgwater, D. C. Elliott, D. Meier and P. de Wild, *J. Anal. Appl. Pyrolysis*, 2010, **88**, 53–72.

532 Y. Zheng, D. Y. Chen and X. F. Zhu, *J. Anal. Appl. Pyrolysis*, 2013, **104**, 514–520.
533 G. Z. Jiang, D. J. Nowakowski and A. V. Bridgwater, *Energy Fuels*, 2010, **24**, 4470–4475.
534 J. M. Cho, S. Chu, P. J. Dauenhauer and G. W. Huber, *Green Chem.*, 2012, **14**, 428–439.
535 Z. Q. Ma, E. Troussard and J. A. van Bokhoven, *Appl. Catal., A*, 2012, **423**, 130–136.
536 Z. Q. Ma, V. Custodis and J. A. van Bokhoven, *Catal. Sci. Technol.*, 2014, **4**, 766–772.
537 V. B. F. Custodis, C. Bahrle, F. Vogel and J. A. van Bokhoven, *J. Anal. Appl. Pyrolysis*, 2015, **115**, 214–223.
538 M. A. Jackson, D. L. Compton and A. A. Boateng, *J. Anal. Appl. Pyrolysis*, 2009, **85**, 226–230.
539 C. A. Mullen and A. A. Boateng, *Fuel Process. Technol.*, 2010, **91**, 1446–1458.
540 M. Zhang and A. Moutsoglou, *Energy Fuels*, 2014, **28**, 1066–1073.
541 M. Zhang, F. L. P. Resende and A. Moutsoglou, *Fuel*, 2014, **116**, 358–369.
542 K.-I. Kuroda, Y. Inoue and K. Sakai, *J. Anal. Appl. Pyrolysis*, 1990, **18**, 59–69.
543 R. Lou, S. B. Wu and G. J. Lyu, *J. Anal. Appl. Pyrolysis*, 2015, **111**, 27–32.
544 B. Iatridis and G. R. Gavalas, *Ind. Eng. Chem. Prod. Res. Dev.*, 1979, **18**, 127–130.
545 Y. Zhao, L. Deng, B. Liao, Y. Fu and Q. X. Guo, *Energy Fuels*, 2010, **24**, 5735–5740.
546 P. J. De Wild, W. J. J. Huijgen and R. J. A. Gosselink, *Biofuels, Bioprod. Biorefin.*, 2014, **8**, 645–657.
547 P. de Wild, R. Van der Laan, A. Kloekhorst and E. Heeres, *Environ. Prog. Sustainable Energy*, 2009, **28**, 461–469.
548 P. J. de Wild, W. J. J. Huijgen and H. J. Heeres, *J. Anal. Appl. Pyrolysis*, 2012, **93**, 95–103.
549 S. Zhou, R. C. Brown and X. L. Bai, *Green Chem.*, 2015, **17**, 4748–4759.
550 W. Kaminsky, W. Schweers and H. Schwesinger, *Holzforschung*, 1980, **34**, 73–75.
551 T. N. Trinh, P. A. Jensen, Z. Sarossy, K. Dam-Johansen, N. O. Knudsen, H. R. Sorensen and H. Egsgaard, *Energy Fuels*, 2013, **27**, 3802–3810.
552 S. Farag, D. B. Fu, P. G. Jessop and J. Chaouki, *J. Anal. Appl. Pyrolysis*, 2014, **109**, 249–257.
553 S. Farag, L. Kouisni and J. Chaouki, *Energy Fuels*, 2014, **28**, 1406–1417.
554 S. Farag, B. P. Mudraboyina, P. G. Jessop and J. Chaouki, *Biomass Bioenergy*, 2016, **95**, 344–353.
555 The reported carbon yields (C%) are converted to mass yields (wt%) by suggesting that softwood kraft lignin is composed of coniferyl alcohol units and by taking toluene and 4-vinylguaiacol as representative products for respectively CFP and fast pyrolysis.
556 D. K. Shen, S. Gu, K. H. Luo, S. R. Wang and M. X. Fang, *Bioresour. Technol.*, 2010, **101**, 6136–6146.
557 R. J. Evans, T. A. Milne and M. N. Soltys, *J. Anal. Appl. Pyrolysis*, 1986, **9**, 207–236.
558 K. Wang, K. H. Kim and R. C. Brown, *Green Chem.*, 2014, **16**, 727–735.
559 X. Li, L. Su, Y. Wang, Y. Yu, C. Wang, X. Li and Z. Wang, *Front. Environ. Sci. Eng.*, 2012, **6**, 295–303.
560 D. J. Mihalcik, C. A. Mullen and A. A. Boateng, *J. Anal. Appl. Pyrolysis*, 2011, **92**, 224–232.
561 Y. Yu, X. Li, L. Su, Y. Zhang, Y. Wang and H. Zhang, *Appl. Catal., A*, 2012, **447–448**, 115–123.
562 Y. Xue, S. Zhou and X. Bai, *ACS Sustainable Chem. Eng.*, 2016, **4**, 4237–4250.
563 J. Q. Bond, A. A. Upadhye, H. Olcay, G. A. Tompsett, J. Jae, R. Xing, D. M. Alonso, D. Wang, T. Zhang, R. Kumar, A. Foster, S. M. Sen, C. T. Maravelias, R. Malina, S. R. H. Barrett, R. Lobo, C. E. Wyman, J. A. Dumesic and G. W. Huber, *Energy Environ. Sci.*, 2014, **7**, 1500–1523.
564 G. F. Zhou, P. A. Jensen, D. M. Le, N. O. Knudsen and A. D. Jensen, *Green Chem.*, 2016, **18**, 1965–1975.
565 H. Ben and A. J. Ragauskas, *ACS Sustainable Chem. Eng.*, 2013, **1**, 316–324.
566 R. Thilakaratne, J.-P. Tessonnier and R. C. Brown, *Green Chem.*, 2016, **18**, 2231–2239.
567 C. Amen-Chen, H. Pakdel and C. Roy, *Bioresour. Technol.*, 2001, **79**, 277–299.
568 E. Dorrestijn, L. J. J. Laarhoven, I. W. C. E. Arends and P. Mulder, *J. Anal. Appl. Pyrolysis*, 2000, **54**, 153–192.
569 M. P. Pandey and C. S. Kim, *Chem. Eng. Technol.*, 2011, **34**, 29–41.
570 S. Kim, S. C. Chmely, M. R. Nimlos, Y. J. Bomble, T. D. Foust, R. S. Paton and G. T. Beckham, *J. Phys. Lett.*, 2011, **2**, 2846–2852.
571 M. V. Galkin, C. Dahlstrand and J. S. M. Samec, *ChemSusChem*, 2015, **8**, 2187–2192.
572 A. Rahimi, A. Azarpira, H. Kim, J. Ralph and S. S. Stahl, *J. Am. Chem. Soc.*, 2013, **135**, 6415–6418.
573 A. Rahimi, A. Ulbrich, J. J. Coon and S. S. Stahl, *Nature*, 2014, **515**, 249–252.
574 J. D. Nguyen, B. S. Matsuura and C. R. J. Stephenson, *J. Am. Chem. Soc.*, 2014, **136**, 1218–1221.
575 N. Luo, M. Wang, H. Li, J. Zhang, H. Liu and F. Wang, *ACS Catal.*, 2016, **6**, 7716–7721.
576 M. Wang, J. M. Lu, X. C. Zhang, L. H. Li, H. J. Li, N. C. Luo and F. Wang, *ACS Catal.*, 2016, **6**, 6086–6090.
577 R. Zhu, B. Wang, M. Cui, J. Deng, X. Li, Y. Ma and Y. Fu, *Green Chem.*, 2016, **18**, 2029–2036.
578 N. D. Patil, S. G. Yao, M. S. Meier, J. K. Mobley and M. Crocker, *Org. Biomol. Chem.*, 2015, **13**, 3243–3254.
579 C. Zhang, H. Li, J. Lu, X. Zhang, K. E. MacArthur, M. Heggen and F. Wang, *ACS Catal.*, 2017, **7**, 3419–3429.
580 J. M. Nichols, L. M. Bishop, R. G. Bergman and J. A. Ellman, *J. Am. Chem. Soc.*, 2010, **132**, 12554–12555.
581 D. M. Miles-Barrett, J. R. D. Montgomery, C. S. Lancefield, D. B. Cordes, A. M. Z. Slawin, T. Lebl, R. Carr and N. J. Westwood, *ACS Sustainable Chem. Eng.*, 2017, **5**, 1831–1839.
582 I. Bosque, G. Magallanes, M. Rigoulet, M. D. Kärkäs and C. R. J. Stephenson, *ACS Cent. Sci.*, 2017, **3**, 621–628.

583 B. Sedai, C. Díaz-Urrutia, R. T. Baker, R. Wu, L. A. P. Silks and S. K. Hanson, *ACS Catal.*, 2013, **3**, 3111–3122.
584 H. Liu, M. Wang, H. Li, N. Luo, S. Xu and F. Wang, *J. Catal.*, 2017, **346**, 170–179.
585 Y. Wang, Q. Wang, J. He and Y. Zhang, *Green Chem.*, 2017, **19**, 3135–3141.
586 A. A. Dwiatmoko, S. Lee, H. C. Ham, J.-W. Choi, D. J. Suh and J.-M. Ha, *ACS Catal.*, 2015, **5**, 433–437.
587 M. M. Hepditch and R. K. Thring, *Can. J. Chem. Eng.*, 1997, **75**, 1108–1114.
588 J. M. Pepper, P. E. T. Baylis and E. Adler, *Can. J. Chem.*, 1959, **37**, 1241–1248.
589 J. F. Stanzione, P. A. Giangiulio, J. M. Sadler, J. J. La Scala and R. P. Wool, *ACS Sustainable Chem. Eng.*, 2013, **1**, 419–426.
590 M. Fache, B. Boutevin and S. Caillol, *Green Chem.*, 2016, **18**, 712–725.
591 Y. Liao, A. Dewaele, D. Verboekend and B. F. Sels, in *Bio-Based Solvents*, ed. F. Jerome and R. Luque, John Wiley & Sons, Ltd, 2017.
592 M. Fache, C. Montérémal, B. Boutevin and S. Caillol, *Eur. Polym. J.*, 2015, **73**, 344–362.
593 A. Deneyer, T. Renders, J. Van Aelst, S. Van den Bosch, D. Gabriëls and B. F. Sels, *Curr. Opin. Chem. Biol.*, 2015, **29**, 40–48.
594 C. Zhao, Y. Kou, A. A. Lemonidou, X. Li and J. A. Lercher, *Angew. Chem., Int. Ed.*, 2009, **48**, 3987–3990.
595 C. Zhao and J. A. Lercher, *ChemCatChem*, 2012, **4**, 64–68.
596 N. Yan, Y. Yuan, R. Dykeman, Y. Kou and P. J. Dyson, *Angew. Chem., Int. Ed.*, 2010, **49**, 5549–5553.
597 W. Zhang, J. Chen, R. Liu, S. Wang, L. Chen and K. Li, *ACS Sustainable Chem. Eng.*, 2014, **2**, 683–691.
598 C. Zhang, J. Xing, L. Song, H. Xin, S. Lin, L. Xing and X. Li, *Catal. Today*, 2014, **234**, 145–152.
599 C. Zhao, D. M. Camaioni and J. A. Lercher, *J. Catal.*, 2012, **288**, 92–103.
600 J. Chen, J. Huang, L. Chen, L. Ma, T. Wang and U. I. Zakai, *ChemCatChem*, 2013, **5**, 1598–1605.
601 C. Zhao and J. A. Lercher, *Angew. Chem.*, 2012, **124**, 6037–6042.
602 C. Zhao, Y. Kou, A. A. Lemonidou, X. Li and J. A. Lercher, *Chem. Commun.*, 2010, **46**, 412–414.
603 C. Zhang, J. Qi, J. Xing, S. Tang, L. Song, Y. Sun, C. Zhang, H. Xin and X. Li, *RSC Adv.*, 2016, **6**, 104398–104406.
604 K. L. Luska, P. Migowski, S. El Sayed and W. Leitner, *Angew. Chem., Int. Ed.*, 2015, **54**, 15750–15755.
605 G.-H. Wang, Z. Cao, D. Gu, N. Pfänder, A.-C. Swertz, B. Spliethoff, H.-J. Bongard, C. Weidenthaler, W. Schmidt, R. Rinaldi and F. Schüth, *Angew. Chem., Int. Ed.*, 2016, **55**, 8850–8855.
606 S. L. Yohe, H. J. Choudhari, D. D. Mehta, P. J. Dietrich, M. D. Detwiler, C. M. Akatay, E. A. Stach, J. T. Miller, W. N. Delgass and R. Agrawal, *J. Catal.*, 2016, **344**, 535–552.
607 H. Ohta, H. Kobayashi, K. Hara and A. Fukuoka, *Chem. Commun.*, 2011, **47**, 12209–12211.
608 T. Prasomsri, M. Shetty, K. Murugappan and Y. Román-Leshkov, *Energy Environ. Sci.*, 2014, **7**, 2660–2669.
609 D. W. Bruce, *Transition Metal Sulphides: Chemistry and Catalysis*, Kluwer Academic Publishers, Dordrecht, 2000.
610 D. J. Rensel, S. Rouvimov, M. E. Gin and J. C. Hicks, *J. Catal.*, 2013, **305**, 256–263.
611 S. Boonyasuwat, T. Omotoso, D. E. Resasco and S. P. Crossley, *Catal. Lett.*, 2013, **143**, 783–791.
612 J. Sun, A. M. Karim, H. Zhang, L. Kovarik, X. S. Li, A. J. Hensley, J.-S. McEwen and Y. Wang, *J. Catal.*, 2013, **306**, 47–57.
613 Y.-B. Huang, L. Yan, M.-Y. Chen, Q.-X. Guo and Y. Fu, *Green Chem.*, 2015, **17**, 3010–3017.
614 H. Ohta, B. Feng, H. Kobayashi, K. Hara and A. Fukuoka, *Catal. Today*, 2014, **234**, 139–144.
615 B. Feng, H. Kobayashi, H. Ohta and A. Fukuoka, *J. Mol. Catal. A: Chem.*, 2014, **388**, 41–46.
616 Z. Luo, Z. Zheng, Y. Wang, G. Sun, H. Jiang and C. Zhao, *Green Chem.*, 2016, **18**, 5845–5858.
617 X. Wang and R. Rinaldi, *Angew. Chem., Int. Ed.*, 2013, **52**, 11499–11503.
618 A. L. Jongerius, R. Jastrzebski, P. C. Bruijnincx and B. M. Weckhuysen, *J. Catal.*, 2012, **285**, 315–323.
619 N. Joshi and A. Lawal, *Ind. Eng. Chem. Res.*, 2013, **52**, 4049–4058.
620 P. Ruiz, B. Frederick, W. De Sisto, R. Austin, L. Radovic, K. Leiva, R. García, N. Escalona and M. Wheeler, *Catal. Commun.*, 2012, **27**, 44–48.
621 T. Omotoso, S. Boonyasuwat and S. P. Crossley, *Green Chem.*, 2014, **16**, 645–652.
622 A. L. Jongerius, R. W. Gosselink, J. Dijkstra, J. H. Bitter, P. C. A. Bruijnincx and B. M. Weckhuysen, *ChemCatChem*, 2013, **5**, 2964–2972.
623 H. Y. Zhao, D. Li, P. Bui and S. T. Oyama, *Appl. Catal., A*, 2011, **391**, 305–310.
624 Z. Cao, J. Engelhardt, M. Dierks, M. T. Clough, G. H. Wang, E. Heracleous, A. Lappas, R. Rinaldi and F. Schüth, *Angew. Chem., Int. Ed.*, 2017, **56**, 2334–2339.
625 Z. Bai, W. C. Phuan, J. Ding, T. H. Heng, J. Luo and Y. Zhu, *ACS Catal.*, 2016, **6**, 6141–6145.
626 W. Schutyser, S. Van den Bosch, J. Dijkmans, S. Turner, M. Meledina, G. Van Tendeloo, D. P. Debecker and B. F. Sels, *ChemSusChem*, 2015, **8**, 1805–1818.
627 W. Schutyser, G. Van den Bossche, A. Raaffels, S. Van den Bosch, S.-F. Koelewijn, T. Renders and B. F. Sels, *ACS Sustainable Chem. Eng.*, 2016, **4**, 5336–5346.
628 X. Wang and R. Rinaldi, *Energy Environ. Sci.*, 2012, **5**, 8244–8260.
629 X. Liu, L. Xu, G. Xu, W. Jia, Y. Ma and Y. Zhang, *ACS Catal.*, 2016, **6**, 7611–7620.
630 G.-Y. Xu, J.-H. Guo, Y.-C. Qu, Y. Zhang, Y. Fu and Q.-X. Guo, *Green Chem.*, 2016, **18**, 5510–5517.
631 M. Ishikawa, M. Tamura, Y. Nakagawa and K. Tomishige, *Appl. Catal., B*, 2016, **182**, 193–203.
632 Y. Nakagawa, M. Ishikawa, M. Tamura and K. Tomishige, *Green Chem.*, 2014, **16**, 2197–2203.
633 D. Verboekend, Y. Liao, W. Schutyser and B. F. Sels, *Green Chem.*, 2016, **18**, 297–306.

634 J. Dijkmans, W. Schutyser, M. Dusselier and B. Sels, *Chem. Commun.*, 2016, **52**, 6712–6715.
635 E. Sanchez-Gonzalez, A. Lopez-Olvera, O. Monroy, J. Aguilar-Pliego, J. Gabriel Flores, A. Islas-Jacome, M. A. Rincon-Guevara, E. Gonzalez-Zamora, B. Rodriguez-Molina and I. A. Ibarra, *CrystEngComm*, 2017, **19**, 4142–4146.
636 R. Delisi, R. Ciriminna, F. Parrino, L. Palmisano, Y.-J. Xu and M. Pagliaro, *ChemistrySelect*, 2016, **1**, 626–629.
637 R. Yepez, S. García, P. Schachat, M. Sánchez-Sánchez, J. H. González-Estefan, E. González-Zamora, I. A. Ibarra and J. Aguilar-Pliego, *New J. Chem.*, 2015, **39**, 5112–5115.
638 M. Fache, E. Darroman, V. Besse, R. Auvergne, S. Caillol and B. Boutevin, *Green Chem.*, 2014, **16**, 1987–1998.
639 J. F. Stanzione Iii, J. M. Sadler, J. J. La Scala, K. H. Reno and R. P. Wool, *Green Chem.*, 2012, **14**, 2346–2352.
640 J. Kayalvizhi and A. Pandurangan, *Mol. Catal.*, 2017, **436**, 67–77.
641 M. Chatterjee, T. Ishizaka and H. Kawanami, *Green Chem.*, 2016, **18**, 487–496.
642 M. Pelckmans, T. Renders, S. Van de Vyver and B. F. Sels, *Green Chem.*, 2017, **19**, 5303–5331.
643 H. Bilel, N. Hamdi, F. Zagrouba, C. Fischmeister and C. Bruneau, *RSC Adv.*, 2012, **2**, 9584–9589.
644 B. G. Harvey, C. M. Sahagun, A. J. Guenthner, T. J. Groshens, L. R. Cambrea, J. T. Reams and J. M. Mabry, *ChemSusChem*, 2014, **7**, 1964–1969.
645 K. A. Alexander, E. A. Paulhus, G. M. L. Lazarus and N. E. Leadbeater, *J. Organomet. Chem.*, 2016, **812**, 74–80.
646 S. F. Koelewijn, S. Van den Bosch, T. Renders, W. Schutyser, B. Lagrain, M. Smet, J. Thomas, W. Dehaen, P. Van Puyvelde, H. Witters and B. F. Sels, *Green Chem.*, 2017, **19**, 2561–2570.
647 H. A. Meylemans, T. J. Groshens and B. G. Harvey, *ChemSusChem*, 2012, **5**, 206–210.
648 P. Ferrini, S.-F. Koelewijn, J. Van Aelst, N. Nuttens and B. F. Sels, *ChemSusChem*, 2017, **10**, 2249–2257.
649 A. Kaiho, D. Mazzarella, M. Satake, M. Kogo, R. Sakai and T. Watanabe, *Green Chem.*, 2016, **18**, 6526–6535.
650 G. Fuchs, M. Boll and J. Heider, *Nat. Rev. Microbiol.*, 2011, **9**, 803–816.
651 R. Y. Stainer and L. N. Ornston, *Adv. Microb. Physiol.*, 1973, **9**, 89–151.
652 L. Ornston and R. Stanier, *J. Biol. Chem.*, 1966, **241**, 3776–3786.
653 C. S. Harwood and R. E. Parales, *Annu. Rev. Microbiol.*, 1996, **50**, 553–590.
654 M. P. McLeod, R. L. Warren, W. W. L. Hsiao, N. Araki, M. Myhre, C. Fernandes, D. Miyazawa, W. Wong, A. L. Lillquist, D. Wang, M. Dosanjh, H. Hara, A. Petrescu, R. D. Morin, G. Yang, J. M. Stott, J. E. Schein, H. Shin, D. Smailus, A. S. Siddiqui, M. A. Marra, S. J. M. Jones, R. Holt, F. S. L. Brinkman, K. Miyauchi, M. Fukuda, J. E. Davies, W. W. Mohn and L. D. Eltis, *Proc. Natl. Acad. Sci. U. S. A.*, 2006, **103**, 15582–15587.
655 J. I. Jiménez, B. Miñambres, J. L. García and E. Díaz, *Environ. Microbiol.*, 2002, **4**, 824–841.
656 K. E. Nelson, C. Weinel, I. T. Paulsen, R. J. Dodson, H. Hilbert, V. A. P. Martins dos Santos, D. E. Fouts, S. R. Gill, M. Pop, M. Holmes, L. Brinkac, M. Beanan, R. T. DeBoy, S. Daugherty, J. Kolonay, R. Madupu, W. Nelson, O. White, J. Peterson, H. Khouri, I. Hance, P. C. Lee, E. Holtzapple, D. Scanlan, K. Tran, A. Moazzez, T. Utterback, M. Rizzo, K. Lee, D. Kosack, D. Moestl, H. Wedler, J. Lauber, D. Stjepandic, J. Hoheisel, M. Straetz, S. Heim, C. Kiewitz, J. Eisen, K. N. Timmis, A. Düsterhöft, B. Tümmler and C. M. Fraser, *Environ. Microbiol.*, 2002, **4**, 799–808.
657 V. Barbe, D. Vallenet, N. Fonknechten, A. Kreimeyer, S. Oztas, L. Labarre, S. Cruveiller, C. Robert, S. Duprat, P. Wincker, L. N. Ornston, J. Weissenbach, P. Marlière, G. N. Cohen and C. Médigue, *Nucleic Acids Res.*, 2004, **32**, 5766–5779.
658 E. Masai, N. Kamimura, D. Kasai, A. Oguchi, A. Ankai, S. Fukui, M. Takahashi, I. Yashiro, H. Sasaki, T. Harada, S. Nakamura, Y. Katano, S. Narita-Yamada, H. Nakazawa, H. Hara, Y. Katayama, M. Fukuda, S. Yamazaki and N. Fujita, *J. Bacteriol.*, 2012, **194**, 534–535.
659 J. D. Lipscomb, *Curr. Opin. Struct. Biol.*, 2008, **18**, 644–649.
660 F. H. Vaillancourt, J. T. Bolin and L. D. Eltis, *Crit. Rev. Biochem. Mol. Biol.*, 2006, **41**, 241–267.
661 S. Harayama, M. Kok and E. Neidle, *Annual Reviews in Microbiology*, 1992, **46**, 565–601.
662 T. D. H. Bugg, *Tetrahedron*, 2003, **59**, 7075–7101.
663 T. D. H. Bugg and R. Rahmanpour, *Curr. Opin. Chem. Biol.*, 2015, **29**, 10–17.
664 G. T. Beckham, C. W. Johnson, E. M. Karp, D. Salvachúa and D. R. Vardon, *Curr. Opin. Biotechnol.*, 2016, **42**, 40–53.
665 O. Y. Abdelaziz, D. P. Brink, J. Prothmann, K. Ravi, M. Sun, J. García-Hidalgo, M. Sandahl, C. P. Hulteberg, C. Turner, G. Lidén and M. F. Gorwa-Grauslund, *Biotechnol. Adv.*, 2016, **34**, 1318–1346.
666 E. Masai, Y. Katayama and M. Fukuda, *Biosci., Biotechnol., Biochem.*, 2007, **71**, 1–15.
667 E. Masai, A. Ichimura, Y. Sato, K. Miyauchi, Y. Katayama and M. Fukuda, *J. Bacteriol.*, 2003, **185**, 1768–1775.
668 T. D. H. Bugg, M. Ahmad, E. M. Hardiman and R. Singh, *Curr. Opin. Biotechnol.*, 2011, **22**, 394–400.
669 T. D. H. Bugg, M. Ahmad, E. M. Hardiman and R. Rahmanpour, *Nat. Prod. Rep.*, 2011, **28**, 1883–1896.
670 A. M. Varman, L. He, R. Follenfant, W. H. Wu, S. Wemmer, S. A. Wrobel, Y. J. J. Tang and S. Singh, *Proc. Natl. Acad. Sci. U. S. A.*, 2016, **113**, E5802–E5811.
671 P. D. Sainsbury, E. M. Hardiman, M. Ahmad, H. Otani, N. Seghezzi, L. D. Eltis and T. D. H. Bugg, *ACS Chem. Biol.*, 2013, **8**, 2151–2156.
672 K. M. Draths and J. W. Frost, *J. Am. Chem. Soc.*, 1994, **116**, 399–400.
673 D. R. Vardon, N. A. Rorrer, D. Salvachua, A. E. Settle, C. W. Johnson, M. J. Menart, N. S. Cleveland, P. N. Ciesielski, K. X. Steirer, J. R. Dorgan and G. T. Beckham, *Green Chem.*, 2016, **18**, 3397–3413.
674 C. W. Johnson, D. Salvachúa, P. Khanna, H. Smith, D. J. Peterson and G. T. Beckham, *Metabolic Engineering Communications*, 2016, **3**, 111–119.
675 C. W. Johnson, P. E. Abraham, J. G. Linger, P. Khanna, R. L. Hettich and G. T. Beckham, *Metabolic Engineering Communications*, 2017, **5**, 19–25.

676 Z. Mycroft, M. Gomis, P. Mines, P. Law and T. D. H. Bugg, *Green Chem.*, 2015, **17**, 4974–4979.
677 C. W. Johnson and G. T. Beckham, *Metab. Eng.*, 2015, **28**, 240–247.
678 D. Salvachua, E. M. Karp, C. T. Nimlos, D. R. Vardon and G. T. Beckham, *Green Chem.*, 2015, **17**, 4951–4967.
679 L. Lin, Y. Cheng, Y. Pu, S. Sun, X. Li, M. Jin, E. A. Pierson, D. C. Gross, B. E. Dale, S. Y. Dai, A. J. Ragauskas and J. S. Yuan, *Green Chem.*, 2016, **18**, 5536–5547.
680 C. Zhao, S. Xie, Y. Pu, R. Zhang, F. Huang, A. J. Ragauskas and J. S. Yuan, *Green Chem.*, 2016, **18**, 1306–1312.
681 S. Xie, Q. Sun, Y. Pu, F. Lin, S. Sun, X. Wang, A. J. Ragauskas and J. S. Yuan, *ACS Sustainable Chem. Eng.*, 2017, **5**, 2215–2223.
682 D. Salvachua, R. Katahira, N. S. Cleveland, P. Khanna, M. G. Resch, B. A. Black, S. O. Purvine, E. M. Zink, A. Prieto, M. J. Martinez, A. T. Martinez, B. A. Simmons, J. M. Gladden and G. T. Beckham, *Green Chem.*, 2016, **18**, 6046–6062.

