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Solid-State NMR Investigations of Cellulose Structure and Interactions with Matrix Polysaccharides in Plant Primary Cell Walls

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Highlight:
This article discusses the use of multidimensional Solid-State NMR to site-specifically detect polysaccharide interactions and the chain numbers of cellulose microfibrils in near-native primary cell wall samples.

Abstract
Until recently, the three-dimensional architecture of plant cell walls was poorly understood due to the lack of high-resolution techniques for characterizing the molecular structure, dynamics and intermolecular interactions of the wall polysaccharides in these insoluble biomolecular mixtures. We introduced multidimensional solid-state nuclear magnetic resonance (SSNMR) spectroscopy, coupled with $^{13}$C labeling of whole plants, to determine the spatial arrangements of macromolecules in near-native plant cell walls. Here we review key evidence from 2D and 3D correlation NMR spectra that show relatively few cellulose-hemicellulose cross peaks but many cellulose-pectin cross peaks, indicating that cellulose microfibrils are not extensively coated by hemicellulose and all three major polysaccharides exist in a single network rather than two separate networks as previously proposed. The number of glucan chains in the primary-wall cellulose microfibrils has been under active debate recently. We show detailed analysis of quantitative $^{13}$C SSNMR spectra of cellulose in various wild-type (WT) and mutant Arabidopsis and Brachypodium primary cell walls, which consistently indicate that primary-wall cellulose microfibrils contain at least 24 glucan chains.
Abbreviations

Arabinose: Ara, A
Galactose: Gal
Galacturonic acid: GalA, GA
Glucuronic acid: GlcA
Glucose in Xyloglucan: G
Glucuronoarabinoxylan: GAX
Fucose: Fuc
Homogalacturonan: HG, HGA
Mixed-linkage glucan: MLG
Interior crystalline cellulose: i
Surface amorphous cellulose: s
Core interior cellulose: c
Surface-bound interior cellulose: b
Rhamnogalacturonan I: RGI
Rhamnose: Rha, R
Xyloglucan: XyG
Xylose: Xyl, x
Solid-State nuclear magnetic resonance: SSNMR
Cross polarization: CP
Direct polarization: DP
Magic-angle spinning: MAS
Proton-driven spin diffusion: PDS
d Dipolar-assisted rotational resonance: DARR
Double-quantum-filtered: DQF
Incredible natural-abundance double-quantum transfer experiment: INADEQUATE
Introduction

Plant primary cell walls contain many macromolecules, including cellulose, hemicelluloses, pectins and glycoproteins. Cellulose microfibrils consist of linear glucan chains that are held together by hydrogen bonds and other non-covalent interactions (Jarvis, 2003; Nishiyama et al., 2002; Nishiyama et al., 2003b). In dicotyledonous plants, the main hemicellulose is xyloglucan (XyG), which are glucan chains substituted with xylose (Xyl), galactose (Gal), and fucose (Fuc) sidechains (Fry, 1989; Park and Cosgrove, 2015). In grass primary walls, the main hemicelluloses are glucuronoarabinoxylan (GAX) and mixed-linkage glucon (MLG). GAX has a β-(1,4)-linked xylose (Xyl) backbone and arabinose (Ara) and glucuronic acid (GlcA) sidechains, while MLGs are unbranched chains of ~30% β-(1,3) and ~70% β-(1,4)-linked glucopyranosyl residues (Kiemle et al., 2014; Woodward et al., 1988). Pectins are acidic polysaccharides rich in galacturonic acid (GalA) residues. In dicot primary cell walls, both linear homogalacturonan (HG) and branched rhamnogalacturonan (RG) with arabinose and galactose (Gal) sidechains are present (Caffall and Mohnen, 2009), while grass primary walls contain only low concentrations of pectins (Vogel, 2008).

Although the chemical structures and compositions of plant cell wall polymers are relatively well known, how these wall polymers form a three-dimensional network to provide mechanical strength to the wall while allowing the wall to expand and grow is still poorly understood (Cosgrove, 2001, 2014). Molecular-level three-dimensional structural information is difficult to obtain because of the insoluble nature of the cell wall and the amorphous nature of most wall polysaccharides except cellulose (Cosgrove, 2005; Jarvis, 1992). Thus, decades of cell wall structure characterization mainly involved chemical extractions followed by sugar analysis and microscopic imaging, which are limited by significant perturbation of the wall structure and insufficient spatial resolution (Mccann et al., 1995; Mccann et al., 1990; Talbott and Ray, 1992). In vitro binding assays have been used to measure the binding affinities between different wall polysaccharides, but they cannot reproduce the complex molecular interactions in the native wall after biosynthesis. VanderHart and Atalla pioneered the use of 13C solid-state NMR (SSNMR) spectroscopy to characterize purified cellulose in higher plants (Atalla and Vanderhart, 1984; Atalla and Vanderhart, 1999). From the 13C chemical shifts they resolved two cellulose allomorphs, Iα and Iβ. Recently, advanced 2D correlation SSNMR techniques were used to definitively assign these 13C chemical shifts and obtain 1H chemical shifts in bacterial and tunicate cellulose (Kono et al., 2003; Kono and Numata, 2006). These data indicate that the anhydroglucose residues in the two cellulose allomorphs have distinct conformations and are distributed differently in the glucan chains. SSNMR has also been used to investigate polysaccharide structures in secondary cell walls (Bardet et al., 1997; Dupree et al., 2015), protein cross linking in soybean cell walls (Cegelski et al., 2010), and effects of hydration on polysaccharide mobility in onion cell walls (Hediger et al., 1999; Hediger et al., 2002). However, these SSNMR studies did not give information on cellulose interactions with matrix polysaccharides in primary cell walls.

Recently, we introduced 2D and 3D correlation SSNMR for investigating the intermolecular interactions of polysaccharides in near-native, hydrated plant primary cell walls (Dick-Perez et al., 2011). By labeling whole plants with 13C, we obtained sufficient sensitivity to conduct multidimensional correlation SSNMR experiments, which are necessary for resolving the signals of multiple wall polysaccharides. In this way, we obtained site-specific information about the conformation, dynamics, water interaction and intermolecular contacts of the macromolecules in near-native plant cell walls. In this paper, we review key results from these multidimensional correlation SSNMR data. We show that there are relatively few cellulose-xyloglucan cross peaks but many cellulose-pectin cross peaks in Arabidopsis cell walls, which revise the conventional “tethered network” model of the cell wall and suggest instead a single cohesive network in which cellulose...
contacts both pectins and xyloglucan (Dick-Perez et al., 2011; Wang et al., 2012). Similarly, we observed cellulose-GAX cross peaks in the cell wall of the model grass Brachypodium, which provide new insight into polysaccharide interactions in grass cell walls. The major findings of these SSNMR studies are summarized in Table 1.

The C4 chemical shifts of most native celluloses exhibit two resolved bands centered at 89 ppm and 85 ppm, which have long been attributed to interior and surface chains of the microfibril, respectively (Earl and VanderHart, 1981). The relative intensities of these surface and interior cellulose peaks were recently used together with X-ray diffraction and computational modeling to constrain the cross-sectional area of cellulose microfibrils in plants, resulting in structural models with as few as 15 chains and as many as 25 chains (Fernandes et al., 2011; Kennedy et al., 2007; Newman et al., 1996; Newman et al., 1994; Newman et al., 2013; Thomas et al., 2013). These estimates are much smaller than the original 36-chain model proposed based on the hypothesized number of cellulose-synthase proteins in the plasma membrane (Guerriero et al., 2010; Scheible et al., 2001; Taylor, 2008), and the 18-chain model was particularly guided by emerging biochemical data indicating the stoichiometry of the different cellulose synthases in hexameric rosettes and computational modeling of the cellulose synthase structure (Hill et al., 2014; Sethaphong et al., 2013). In this paper, we provide the first quantitative analysis of the intensities of interior and surface cellulose C4 signals in several plant primary cell walls. The resulting, more accurate, intensity ratios indicate a minimum number of 24 chains in both dicot and grass primary cell-wall cellulose microfibrils.

**Plant cell wall $^{13}$C labeling for magic-angle-spinning SSNMR**

The main requirement for 2D and 3D $^{13}$C magic-angle-spinning (MAS) solid-state NMR studies of plant cell walls is $^{13}$C enrichment of the cell wall. This $^{13}$C labeling gives the necessary sensitivity to correlate and resolve the signals of many polysaccharides and proteins. We labeled Arabidopsis and Brachypodium primary cell walls by growing the plants in liquid culture containing $^{13}$C-labeled glucose in the dark. By restricting the growth period to two weeks, we produced chiefly primary cell walls with negligible amounts of secondary cell walls, as confirmed by the lack of lignin signals in the SSNMR spectra (Dick-Perez et al., 2011). Whole seedlings were harvested and intracellular molecules and starch were removed by sodium dodecyl sulfate, sodium metabisulfate, and amylase. All cell wall samples for these SSNMR studies were well hydrated (40-80 wt% water): the first samples involved freeze-drying of the wall followed by rehydration (Dick-Perez et al., 2011; Wang et al., 2012), while subsequent samples were never dried (Wang et al., 2014; White et al., 2014). Our recent comparison of the rehydrated and never-dried cell walls found that polysaccharides $^{13}$C chemical shifts, nuclear-spin relaxation times, and intermolecular cross peaks are indistinguishable (Wang et al., 2015a), indicating that the molecular-level structure and dynamics of wall polysaccharides are reproducible and reversible upon rehydration. Comparison of the quantitative NMR spectra with sugar analysis results (Dick-Perez et al., 2011; White et al., 2014) also indicates that the $^{13}$C labeling is relatively uniform for all wall polysaccharides.

$^{13}$C resonance assignment and the nature of cross-peaks in multidimensional SSNMR spectra

The first step in extracting information from the 2D and 3D SSNMR spectra is to resolve and assign the $^{13}$C chemical shifts of the polysaccharides and proteins. We achieved this using a combination of four correlation NMR techniques: 1) 2D $^{13}$C-$^{13}$C double-quantum to single-quantum correlation mediated by through-bond $^{13}$C-$^{13}$C J-coupling. This experiment is called J-INADEQUATE (Bax et al., 1980; Lesage et al., 1997); 2) 2D $^{13}$C-$^{13}$C $^1$H-driven spin diffusion experiments called PSDS or DARR (Takegoshi et al., 2001); 3) Double-quantum-filtered (DQF) 2D $^{13}$C-$^{13}$C correlation experiment using homonuclear dipolar recoupling sequences (Hohwy et al., 1999); 4) 3D $^{13}$C-$^{13}$C-$^{13}$C correlation experiment mediated by $^{13}$C spin diffusion (Li et al., 2010). The J-INADEQUATE
experiment exhibits cross peaks only between bonded $^{13}$C spins, because the polarization is transferred via the electrons in the covalent bond, while the other three experiments exhibit cross peaks between carbons that are close in space, because polarization transfer is mediated by distance-dependent dipolar couplings. Thus, cross peaks from experiments 2) to 4) can occur between directly bonded or non-bonded carbons. For carbons in a uniformly $^{13}$C-labeled sugar residue, relayed dipolar transfer through multiple bonds is highly efficient. Thus, a C1-C3 cross peak, for example, is most likely mediated by relayed C1-C2 and C2-C3 dipolar transfer instead of direct C1-C3 dipolar transfer. The DQF correlation experiment differs from the spin diffusion experiments in that the former mostly exhibits one-bond and two-bond cross peaks, so that the spectra are relatively simple, while the latter can exhibit cross peaks between more distant carbons. To detect long-range $^{13}$C-$^{13}$C distances up to 1 nm, one can increase the spin diffusion mixing time. The intensity buildup of cross peaks with the mixing time contains semi-quantitative information about internuclear distances. To better resolve long-range cross peaks with high structural content from short-range cross peaks that are mainly useful for resonance assignment, we introduced a relaxation-compensated PDSD technique, which produces a difference spectrum that shows only intermolecular cross peaks (Wang et al., 2015b). Further development of SSNMR methods will be desirable for resolving and detecting intermolecular contacts between different wall polymers.

These 2D and 3D $^{13}$C correlation NMR spectra allowed us to type-assign most $^{13}$C signals of the common monosaccharides, namely Glc, Ara, GalA, GlcA, Xyl, rhamnose (Rha) and Gal, in the primary walls of Arabidopsis (Dick-Perez et al., 2011), Brachypodium (Wang et al., 2014) and maize (unpublished data). For the same type of monosaccharide, the $^{13}$C chemical shifts can vary due to different sugar conformations, linkages and hydrogen-bonding patterns. For example, nine types of arabinose signals were resolved in Brachypodium cell walls, which can be assigned to different arabinose linkages in GAX and arabinan (Wang et al., 2014). So far, the resolved polysaccharide-specific signals include all six glucose carbons of interior and surface cellulose (Wang et al., 2012), all five $^{13}$C signals of Xyl in XyG, all Ara signals of arabinan (Dick-Perez et al., 2011), and all signals of Xyl, GlcA and ferulic acid in GAX (Wang et al., 2014). The Glc backbone and Gal sidechain of XyG are incompletely resolved due to signal overlap from surface cellulose and galactan, respectively. In addition, GalA signals are often used to denote pectin backbones, but their originating polysaccharides, HG and RG, cannot yet be distinguished. Polysaccharide-specific isotopic labeling will be useful to further resolve these signals.

Higher magnetic fields significantly enhance the resolution of the cell wall NMR spectra. To date, the highest field strength we have used for plant cell walls is 21.1 Tesla, corresponding to a $^1$H Larmor frequency of 900 MHz. At this field strength, at least two types of crystalline cellulose signals have been observed (Wang et al., 2012) and dramatic improvement of spectral resolution is seen for matrix polysaccharides. For example, Fig. 1 compares the 2D J-INADEQUATE spectra of the same Arabidopsis primary cell wall sample at 400, 600, and 900 MHz. The $^{13}$C linewidths of matrix polysaccharides are 0.7-1.4 ppm at 400 MHz, 0.5-1.1 ppm at 600 MHz, and only 0.2-0.5 ppm at 900 MHz. The cellulose spectral resolution is also substantially improved: the linewidths are ~2 ppm at low fields but narrow to 0.7-1.0 ppm at 900 MHz. This line narrowing indicates that the $^{13}$C linewidths of uniformly $^{13}$C-labeled cell walls have a substantial homogeneous contribution due to residual dipolar couplings to $^1$H and $^{13}$C-$^{13}$C J-couplings, which becomes less important at higher magnetic fields. Using the C1 region of the 2D J-INADEQUATE spectra as an example, the 900 MHz spectrum resolves at least 14 peaks while the 400 MHz spectrum resolves only ~10 peaks (Fig. 1). In addition, the 900 MHz spectrum resolves three types of AC2 and four types of AC4, while the spectra measured at lower fields only exhibit one broad AC2 peak and one or two AC4 peaks, partially overlapped with
cellulose signals. With the enhanced resolution at high fields, we can unambiguously resolve signals that are only 0.2-0.3 ppm apart. Further increase of the NMR field strengths to 1.0 GHz and beyond is expected to provide even more benefit for obtaining finer structural details of wall polysaccharides.

The challenge of resolving the polysaccharide signals of native cell walls is also met by exploiting the mobility difference between cellulose and matrix polysaccharides: cellulose is largely immobilized except for the C6 hydroxymethyl group, whereas pectins and hemicellulose are highly mobile with C-H bond order parameters of \( \sim 0.5 \) (Dick-Perez et al., 2011; Wang et al., 2014). This mobility difference allows us to selectively detect the signals of rigid or mobile polysaccharides in separate spectra. For example, the mobile GAX in the Brachypodium primary wall were selectively detected in the 2D J-INADEQUATE spectra measured with direct polarization (DP), and the large number of narrow \(^{13}\)C signals has been assigned to five different Xyl and nine different Ara types, indicating the diverse linkages and substitution patterns of GAX in grass primary walls (Wang et al., 2014). \(^{13}\)C-\(^{1}\)H dipolar dephasing has also been used to suppress the signals of rigid cellulose and detect only those of mobile matrix polysaccharides (Komatsu and Kikuchi, 2013). Conversely, by using short \(^{1}\)H-\(^{13}\)C cross polarization (CP) transfer, we have obtained \(^{13}\)C spectra exhibiting only cellulose signals in the Brachypodium cell wall.

**Intermolecular cross peaks of primary cell wall polysaccharides**

The assignment of most polysaccharide \(^{13}\)C chemical shifts allowed us to determine \(^{13}\)C-\(^{13}\)C cross peaks that reflect intermolecular proximities. With mixing times of 1.5 s and shorter, a conservative estimate of the upper bound of \(^{13}\)C-\(^{13}\)C distances is 10 Å. For the Arabidopsis cell wall, 3D and 2D spectra have been measured with spin diffusion mixing times of up to 300 ms and 1.5 s, respectively (Dick-Perez et al., 2011; Wang et al., 2012). These spectra yielded a number of unambiguous cross peaks between cellulose and pectins, for example, between the crystalline cellulose C4 chemical shifts of 89 ppm and the pectin chemical shifts of 101 ppm and 80 ppm (Fig. 2a). On the other hand, although hemicellulose was long thought to cover the surfaces of cellulose microfibrils, cross peaks between the two are few and ambiguous. The 3D spectra of the Arabidopsis wall showed a few cellulose cross peaks with the XyG backbone Glc and with Gal sidechains (Dick-Perez et al., 2011), but no unambiguous signals between cellulose and xylose were detected. Further experiments that better resolve XyG signals will be useful for verifying the paucity of cellulose-hemicellulose cross peaks. However, the fact that even with partial resonance overlap, such cellulose-hemicellulose cross peaks are not abundant strongly suggests that XyG does not extensively coat the microfibril surface (Boetten et al., 2004). Since in-vitro assembly data showed that XyG has a stronger affinity for cellulose than pectins, these SSNMR data imply that in-vivo wall assembly is quite different from in-vitro assembly, and pectins and hemicellulose may compete for cellulose binding sites in ways that are not replicated in vitro (Wang et al., 2015a).

It is noteworthy that most cellulose-pectin cross peaks observed in the intact cell wall are retained in a sample in which \( \sim 40\% \) of HG had been extracted (Fig. 2a, b) (Wang et al., 2015a). This means that the cellulose-pectin spatial contacts are not due to molecular crowding; rather, RG-I and some of the HG are responsible for contacting cellulose. These findings are consistent with the observation that cellulose-pectin cross peaks are also independent of hydration and temperature, and together suggest that the interactions between pectins and cellulose are specific, and some pectins may be entrapped inside or between the microfibrils (Wang et al., 2015a). Therefore, pectins may play more important roles in wall biomechanics than depicted in the traditional tethered-network model. Indeed, recent biochemical data showed that arabinins and galactans interact strongly with cellulose (Zykwinska et al., 2007), and XyG-deficient cell walls exhibit almost normal development as wild-type cell wall (Cavalier et al., 2008).
The matrix polysaccharides of grass primary cell walls differ chemically from those of dicot primary walls (Carpita, 1996; Carpita and Gibeaut, 1993). In the two-week-old *Brachypodium* primary walls, the main matrix polysaccharide is highly branched GAX (Wang et al., 2014), and no MLG signals were detected. For this two-component cell wall, 2D $^{13}$C correlation spectra exhibited many cellulose-GAX cross peaks, for example between Ara C1 (108.4 ppm) and interior cellulose C4 (88.3 ppm), and between Xyl C1 (102.0 ppm) and interior cellulose C4 (Fig. 2c). Although such GAX-cellulose spatial contact may not seem surprising given the fact that few other matrix polysaccharides are present, the data counter the conventional model that highly branched GAX chains cannot bind cellulose. An implication of this finding is that the cellulose microfibril has sufficient unevenness and surface disorder to accommodate the branched polysaccharides. Again, in vitro binding assays report only a small fraction (4-15%) of GAX binding to cellulose (Carpita, 1983), similar to the low-level of pectin-cellulose binding in vitro, but these results may systematically underestimate the in-vivo intermolecular interactions in the wall.

**Percentages of sugar residues at intermolecular interfaces**

Since the cross-peak intensities at long mixing times reflect the percentage of a polysaccharide in nanometer contact with each other, we can estimate the percentages of sugar residues at intermolecular interfaces. Since each surface cellulose chain must be adjacent to one interior cellulose chain, the surface-interior cellulose cross-peak intensity in the 2D spectra serves as an internal control of the extent of intermolecular contacts between matrix polysaccharides and cellulose. We found that 25-50% of surface cellulose contacts pectins (Wang et al., 2012). This is a very significant percentage not predicted by existing cell-wall structural models. The extent of cellulose-XyG interaction cannot be accurately estimated because of insufficient resolution of the XyG backbone signals in the spectra.

A second approach for estimating the percentages of pectins and XyG that interact with cellulose is by detecting heterogeneous mobilities of the matrix polysaccharides. In both *Arabidopsis* and *Brachypodium* primary walls, cellulose backbone exhibits single-exponential decays for both $^{13}$C spin-lattice ($T_1$) relaxation and $^1$H rotating-frame spin-lattice relaxation (T$_{1\rho}$) (Dick-Perez et al., 2011; Wang et al., 2014), indicating that cellulose is uniformly rigid. In comparison, XyG and pectins in *Arabidopsis* show double-exponential relaxation where 40-60% of a highly mobile component coexists with a rigid component. The most likely interpretation of this bimodal dynamics is that two domains exist in each matrix polysaccharide: the rigid domain interacts with cellulose through van der Waals interaction, hydrogen bonding, or entrapment, while the mobile domain occupies the inter-fibrillar space. Interestingly, in both *Arabidopsis* and *Brachypodium* primary walls, the well resolved 65-ppm peak of interior cellulose C6 also exhibits bimodal relaxation, with the mobile component accounting for ~20% of the total intensity. This mobility could be explained by the freedom of C6 to rotate and change the C4-C5-C6-O6 torsion angle (Fernandes et al., 2011; Matthews et al., 2006) or by the flexibilities of the matrix polysaccharides that contact cellulose, which may influence the exposed C6 more than the embedded ring carbons.

**Single-network model of plant primary walls**

The intermolecular cross peaks in the 2D and 3D $^{13}$C correlation spectra support a single network model of primary cell walls, in which both pectins and hemicellulose interact with cellulose microfibrils. This conclusion is supported by a recent hydration study that found that removal of Ca$^{2+}$ ions that crosslink HG slowed down water $^1$H spin diffusion to both pectins and cellulose (White et al., 2014), indicating that cellulose interacts intimately with pectins. This structural conclusion also found support from recent biomechanical assays showing that the majority of XyG does not have load-
bearing function, since endoglucanases that hydrolyze only XyG or only cellulose do not cause wall 
creep. Instead, an endoglucanase that simultaneously cuts XyG and cellulose loosens the wall, thus 
only a small fraction of XyG binds cellulose as load-bearing tethers (Park and Cosgrove, 2012a, b). 
Intriguingly, these cellulose-XyG “biomechanical hotspots” have been recently found to be the site of 
expansin binding using $^{13}$C spin diffusion NMR (Wang et al., 2013).

**Lateral heterogeneity of cellulose conformations in the microfibril from 2D SSNMR spectra**

Multidimensional $^{13}$C SSNMR not only provides information on the three-dimensional 
arhitecture of the cell wall, but also constrains the cross-sectional area of cellulose microfibrils. The 
C1, C4, and C6 chemical shifts of cellulose have long been known to be diagnostic of cellulose 
crystallinity and allomorphs (Atalla and Vanderhart, 1984; Atalla and Vanderhart, 1999; Horii et al., 
1987). For our analysis below, we assign the C4 and C6 chemical shifts of 89 and 65 ppm to interior 
crystalline glucan chains and the 85 and 62 ppm peaks to surface chains with partial disorder. The 
89/65 ppm interior glucan signals are well resolved from all other polysaccharides’ signals, thus they 
are unambiguous indicators of cellulose. The possibility that the 85/62 ppm chains may reside inside 
the microfibril instead of on the surface is considered low, because the 85/62 ppm peaks have strong 
peak cross with matrix polysaccharides and water (Fernandes et al., 2011; Wang et al., 2012; White et 
al., 2014) and exhibit large-amplitude dynamics (Dick-Perez et al., 2011; Wang et al., 2014). 
Longitudinal disorder of interior glucan chains has been estimated at only 4-5 residues for every 300 
residues (Nishiyama et al., 2003a), thus it should not significantly affect the extracted ratio of surface : 
interior chain numbers.

**Fig. 3** shows the 2D $^{13}$C-$^{13}$C PDSD spectrum of never-dried *Brachypodium* cell walls at 20˚C 
(Wang et al., 2014). The $^{13}$C magnetization was created using a short $^1$H-$^{13}$C CP contact time of 35 μs, 
which suppressed the signals of mobile polysaccharides and gave a predominantly cellulose spectrum 
in the indirect dimension. A long $^{13}$C spin diffusion mixing time of 3.0 s was applied to transfer the $^{13}$C 
magnetization to polysaccharides within ~1 nm of the cellulose. Interestingly, despite the long mixing 
time, the $^{13}$C cross sections of interior and surface cellulose are not identical (**Fig. 3b**), with the 
difference spectrum corresponding to that of pure crystalline cellulose. Long-mixing-time PDSD 
spectra were also measured at low temperature (-20˚C) to freeze molecular motion and with regular CP 
contact times to detect all polysaccharide signals. The resulting surface and interior cellulose cross 
sections still retain their different intensity distributions (Wang et al., 2014). These results indicate that 
some interior glucan chains are separated from the surface chains by more than the distance reach of 
$^{13}$C spin diffusion. Thus, there are two types of interior cellulose chains: a core (c) fraction that is not 
in direct contact with the surface, and a bound (b) fraction that is (**Fig. 3c**). This result dovetails an 
a earlier structural model based on spectral deconvolution, which suggested the presence of a para-
crystalline layer between the microfibril surface and the crystalline core (Larsson et al., 1999). The C6 
of the two interior cellulose fractions resonates at slightly different chemical shifts, 65.5 ppm for the 
core cellulose and 64.8 ppm for the surface-bound interior cellulose (**Fig. 3d**), suggesting that the 
hydroxymethyl conformation depends on the location of the interior chains, with the core cellulose C6 
chemical shift corresponding to that of a trans-gauche (tg) conformer (Vietor et al., 2002). The 
*Brachypodium* result is reproduced in the *Arabidopsis* cell wall (**Fig. 4**), which also exhibits different 
surface and interior cellulose cross sections at long mixing times, with the difference spectrum 
corresponding to the signals of crystalline cellulose. Therefore, cellulose microfibrils in both grass and 
dicot primary walls are sufficiently large to contain three layers of glucan chains.

The core cellulose has two resolved cC1 peaks at 105.5 and 104.0 ppm (**Table 2**), which 
resemble the C1 chemical shifts of I$_{\beta}$ cellulose (Kono et al., 2003). The C3, C5 and C6 chemical shifts
of core cellulose are also similar to those of Iβ cellulose. However, no doublet is observed for C6, as expected for Iβ cellulose. We attribute this absence to insufficient resolution since the two Iβ C6 chemical shifts differ by only 0.6 ppm based on tunicate cellulose data (Table 2) (Kono et al., 2003; Kono and Numata, 2006). The Iβ allomorph contains two types of magnetically inequivalent anhydroglucose residues, which are not directly linked in the same chain but are located in different chains (Kono and Numata, 2006) and perhaps even in alternating sheets (Jarvis, 2003; Nishiyama et al., 2002). It is well known that the Iα allomorph dominates in bacterial and algae while the Iβ allomorph dominates in the secondary cell walls of higher plants (Atalla and Vanderhart, 1984). The iC4 chemical shifts of Arabidopsis primary walls suggest that both Iα and Iβ allomorphs are present (Newman et al., 1996), with Iβ being slightly more abundant. More detailed structural information of the primary-wall cellulose will require more advanced experiments that resolve the 13C chemical shifts of surface cellulose and matrix polysaccharides and that relate 13C chemical shifts to direct conformational parameters such as torsion angles and distances.

**The number of glucan chains in cellulose microfibrils from quantitative 13C SSNMR spectra**

The number of glucan chains in plant cellulose microfibrils has been estimated from the relative intensities of surface and interior cellulose C4 peaks in the solid-state NMR spectra (Kennedy et al., 2007; Newman et al., 1996; Newman et al., 1994). Since 13C spectra also contain matrix polysaccharide signals that partly overlap with the surface cellulose peaks, Newman and coworkers used nuclear-spin relaxation to edit the 13C spectra: linear combinations of CP spectra with and without relaxation filters resulted in predominantly cellulose or predominantly matrix polysaccharide sub-spectra. The cellulose sub-spectrum indicated a crystallinity of 0.37-0.44, which translates to a surface to interior chain-number ratio (s : i) of 1.3-1.7. This range corresponds to an average number of 23 chains in the microfibril (Newman et al., 1996; Newman et al., 1994).

Two assumptions in this relaxation-filtered NMR approach are that surface cellulose has the same dynamic property as interior cellulose and that matrix polysaccharides are fully removed by the relaxation filters due to their fast dynamics. However, recent measurements of spin-diffusion-free 1H T1p relaxation times showed that in hydrated primary cell walls, the surface cellulose is more mobile than interior cellulose, while a non-negligible fraction of matrix polysaccharides is relatively rigid, presumably due to their contact with the cellulose microfibril (Dick-Perez et al., 2011; Wang et al., 2012). Thus, the signals of the rigid fraction of matrix polysaccharides may be difficult to suppress completely in the CP spectra. As a result, the relaxation-filtered 13C spectra may neither represent only the cellulose signals nor capture all cellulose intensities. In addition, 13C CP spectra are inherently non-quantitative unless specially designed pulse sequences are used (Johnson and Schmidt-Rohr, 2014), because the CP process is affected by motion and nuclear spin relaxation, and generally favor the detection of rigid molecules while under-representing dynamic polysaccharides.

Quantitative intensities of surface and interior cellulose are most reliably obtained from 13C direct-polarization (DP) spectra measured with long recycle delays. We measured and compared such quantitative 13C spectra of several plants using recycle delays of 15 - 25 s (Fig. 5), which are sufficiently long to equilibrate the 13C magnetization of these uniformly 13C-labeled cell walls, whose T1 relaxation times have been measured to be 1 - 4 s (Dick-Perez et al., 2011; Wang et al., 2014). These 13C T1 values are much shorter than those of unlabeled cell walls because 13C spin diffusion in the labeled samples is much more efficient and equilibrates the short T1’s of dynamic functional groups with the long T1’s of rigid functional groups. In comparison, the majority of the plant cell wall SSNMR literature involved unlabeled cell wall samples with much longer 13C T1 relaxation times, thus the quantitative 13C DP experiment was not conducted due to its prohibitively low sensitivity, and most

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SSNMR analysis of the surface and interior glucan chain numbers relied on non-quantitative CP spectra.

In total, we examined the $^{13}$C DP spectra of two grass cell walls and five *Arabidopsis* cell walls. Most cell walls were never dried during preparation, except for the xxt1xxt2xxt5 mutant of *Arabidopsis* and samples prepared at the University of Kentucky, which were rehydrated samples (Table 3) (Dick-Perez et al., 2011; Harris et al., 2012; White et al., 2014). We use the 86.8-80.4 ppm range to represent the surface cellulose C4 and unresolved matrix polysaccharide $^{13}$C signals, the 92.0-86.8 ppm range to represent the interior cellulose C4 intensity, and the 111.8-107.2 ppm range to represent the Ara C1 intensity. The *Brachypodium* cell wall contains negligible amounts of XyG, thus the intensity analysis is straightforward. We integrated the interior cellulose C4 (iC4) peak, the Ara C1 peak, and the mixed peak of surface cellulose C4 (sC4), Ara C2 and C4 (Fig. 5a). Since the resolved Ara C1 peak indicates the intensity of a single carbon in Ara, subtraction of twice this intensity from the 86.8-80.4 ppm band yields the intensity of pure surface cellulose C4. In this way, we obtained an $s : i$ ratio of 1.4 for *Brachypodium* cellulose. Similarly, an $s : i$ ratio of 1.3 was found for the *Poa annua* cellulose (Brabham et al., 2014). Spectral deconvolution based on the chemical shifts resolved in 2D $^{13}$C-$^{13}$C correlation spectra yielded very similar $s : i$ ratios with an experimental uncertainty of ±0.1.

For wild-type *Arabidopsis*, the surface cellulose intensities require more care to quantify because of the significant amount of XyG in the wall. We first examined the spectrum of the XyG-depleted xxt1xxt2xxt5 mutant. The $s : i$ ratio was found to be 1.2, in good agreement with the ratios of the grass cell walls. For wild-type *Arabidopsis* walls, the XyG backbone glucose C4 and the surface cellulose C4 signals are unresolved, thus we report the $(s + G) : i$ ratio. Intact walls prepared at Penn State University and the University of Kentucky gave $(s + G) : i$ ratios of 1.8 - 1.9 (Fig. 5c, d). When the majority of matrix polysaccharides were digested by sequential treatments with CDTA, Na$_2$CO$_3$, XEG, Cel12A and 1 M NaOH (White et al., 2014), the intensity ratio decreased to 1.5. Since residual matrix polysaccharides are still present in this digested sample, this value is an upper bound to the $s : i$ ratios in intact wild-type *Arabidopsis* walls. Taken together, the $s : i$ ratios of both grass and dicot primary walls (Table 3) span a range of 1.2 – 1.5, with an estimated error bar of ±0.1. For the reverse-engineered *Arabidopsis* mutant cesal$_{legens}$/cesa3$^{2r1-2}$, a much larger $(s + G) : i$ value of 2.5 was found. This was attributed to the significantly higher percentages of matrix polysaccharides in this mutant plant in response to the reduced crystallinity of cellulose (Harris et al., 2012).

In modeling the number of glucan chains in the microfibril based on these quantitative $s : i$ ratios, we assume that the number of glucan chains in adjacent planes varies by one and the chain numbers are symmetric with respect to the center of the microfibril (Fig. 6). Smaller microfibrils have larger $s : i$ ratios (Supplementary Fig. S1) but models with different numbers of chains sometimes have similar $s : i$ ratios due to different packing geometries (Supplementary Fig. S2). For $s : i$ ratios of 1.2 – 1.5, we found chain numbers of 36 - 24. If we use an $s : i$ ratio of 1.3 as the average value for primary-wall cellulose, then the average number of glucan chains is 28. If we impose the constraint that the chain number is an integer multiples of 6 due to the hexameric structure of cellulose synthase (Endler and Persson, 2011; Herth, 1983), then the most likely chain numbers are 24 and 30. However, irregular microfibril cross sections with other chain numbers should be considered possible at this point. In comparison, small microfibril models with 18 or fewer chains correspond to $s : i$ ratios of larger than 2.0, which deviates from the measured data well beyond the experimental uncertainty, and thus can be excluded. Fig. 6b also shows that small microfibrils with 18 chains or fewer do not have a core domain, instead all interior chains contact the surface chains, which is inconsistent with the long-mixing-time 2D spectra shown in Fig. 3 and Fig. 4. Thus, both the quantitative $^{13}$C spectra and the 2D
PDSD spectra indicate that the cellulose microfibrils in plant primary walls must be sufficiently large to contain at least 24 chains.

Transmission electron microscopy, atomic force microscopy, X-ray scattering, and SSNMR data of plant primary walls generally indicate that the lateral dimension of the cellulose microfibrils is 2-5 nm (Guerriero et al., 2010). In terms of the number of glucan chains, the earliest proposal of 36-chains based on the hypothesized number of cellulose synthase subunits in the rosette structure is now widely considered an over-estimate. Instead, recent proposals have shifted to the other extreme of very small microfibrils containing only 18 chains. The most influential study was based on a joint analysis of wide-angle X-ray scattering (WAXS) and SSNMR data of mung bean cell walls (Newman et al., 2013), in which the WAXS data was deconvoluted using computer-simulated diffractograms of various cellulose models containing 18, 24 or 36 chains with different disorder. These models were simultaneously constrained by the NMR-derived crystallinity factor, X, which is directly related to the s : i ratio. The joint analysis suggested that a mixture of 18-chain microfibrils with irregular shapes and twinning best reproduced the WAXS and SSNMR data. It is of interest to assess the uncertainties in this analysis. First, the cross section shape factor, K, used in calculating the number of (200) planes in the microfibril is assumed to be 0.9 but can vary from 0.84 to 1.0. Using a higher K would increase the number of (200) planes and hence the number of chains. Second, the SSNMR constraint of X = 0.37 corresponds to a large s : i ratio of 1.8, which is inconsistent with the quantitative s : i ratios found here. This large X most likely results from incomplete subtraction of the matrix polysaccharide intensities from the 87-80 ppm band. If s : i ratios of 1.2-1.5 were used (X: 0.45-0.40), and the K value is allowed to vary from 0.9 to 1.0, then the number of chains increases to 20-25, in good agreement with the current analysis. Indeed, the 2013 study pointed out that both the WAXS and SSNMR data can be fit with a 24-chain model if twinning is absent. The 18-chain model fits the s : i ratio of 1.8 only if at least 40% of the cellulose microfibrils twinned and all the chains on the twinning interface are converted to highly crystalline structures so that their C4 and C6 signals would resonate at 89 and 65 ppm. This crystallization process would require the establishment of many hydrogen bonds and likely conformational changes of the hydroxymethyl group. To our knowledge, these two requirements, a high degree of twinning in primary walls and the crystallization of surface chains upon twinning, have not been observed experimentally, thus cautioning against the interpretation of the 18-chain model.

The quantitative s : i ratios obtained from these SSNMR spectra place important constraints on the cellulose structural model. Our findings that some interior chains are more than one chain away from the nearest surface chains, together with the reduced s : i ratios of 1.2-1.5, both indicate that cellulose microfibrils in both dicot and grass primary walls should have sufficiently large dimensions to contain at least 24 chains.

**Conclusions**

Multidimensional $^{13}$C solid-state NMR of $^{13}$C-labeled plants is a powerful and versatile tool to elucidate the spatial proximities and structures of polysaccharides and proteins in near-native plant cell walls. Intermolecular cross peaks indicate that the primary wall of higher plants consists of a single cohesive network of polysaccharides, in which cellulose interacts with both hemicellulose and pectins on the nanometer scale. 2D $^{13}$C-$^{13}$C correlation spectra and 1D quantitative $^{13}$C NMR spectra of dicot and grass primary walls indicate that cellulose microfibrils contain at least 24 glucan chains. This size is sufficiently large for some of the interior chains to avoid direct contact with the surface chains, thus explaining the lack of intensity equilibration between the interior and surface cellulose $^{13}$C signals at long spin diffusion mixing times. Future development of high-resolution SSNMR techniques and the synergistic use of multiple techniques should lead to higher-resolution structure of the cellulose microfibrils and their assemblies.
Supplementary Data

Supplementary data are available at JXB online.
Supplementary Fig. S1. Relationship of the chain number and s : i ratio of cellulose microfibrils.
Supplementary Fig. S2. Cellulose microfibril models with various chain numbers and s : i ratios.

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References


<table>
<thead>
<tr>
<th>Major Findings</th>
<th>Key Experiments</th>
<th>References</th>
</tr>
</thead>
<tbody>
<tr>
<td>There are limited XyG-cellulose contacts but extensive pectin-cellulose contacts in <em>Arabidopsis</em> cell walls.</td>
<td>DQF, INADEQUATE, DIPSHIFT, 3D CCC</td>
<td>(Dick-Perez <em>et al.</em>, 2011)</td>
</tr>
<tr>
<td>Partial depectination rigidifies the remaining wall polymers. 25-50% of the cellulose surface is surrounded by pectins.</td>
<td>PDSD, $^{13}$C-T$<em>1$, $^1$H-T$</em>{lp}$ PDSD buildup analysis, RFDR</td>
<td>(Dick-Perez <em>et al.</em>, 2012)</td>
</tr>
<tr>
<td>Cellulose crystallinity is reduced in <em>cesa1</em>&lt;sup&gt;ageans&lt;/sup&gt;/*cesa3&lt;/sup&gt;xr&lt;sup&gt;1-2&lt;/sup&gt; mutant. Expansin binds XyG-enriched regions of cellulose microfibrils to loosen the cell wall.</td>
<td>CP, DP, INADEQUATE Dynamic Nuclear Polarization, Protein-edited spin diffusion Short-CP PDSD, $^{13}$C-T$<em>1$, $^1$H-T$</em>{lp}$ Water-polysaccharide spin diffusion T$_1$-compensated PDSD</td>
<td>(Harris <em>et al.</em>, 2012) (Wang <em>et al.</em>, 2013)</td>
</tr>
<tr>
<td>GAX and cellulose have sub-nanometer spatial contacts in <em>Brachypodium</em> cell walls. Use water to probe the structure of intact and digested walls.</td>
<td>$^{13}$C-$^1$H MELODI-HETCOR</td>
<td>(Wang <em>et al.</em>, 2014) (White <em>et al.</em>, 2014)</td>
</tr>
<tr>
<td>Intermolecular cross peaks can be selectively detected in a new 2D $^{13}$C correlation experiments. Cellulose-pectin spatial contacts are inherent the primary walls, independent of the hydration history. Never-dried and rehydrated walls show the same cellulose-pectin cross peaks.</td>
<td></td>
<td>(Wang <em>et al.</em>, 2015b) (Wang <em>et al.</em>, 2015a)</td>
</tr>
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Table 2. Cellulose chemical shifts observed in 1D and 2D $^{13}$C SSNMR spectra here and in the literature by multidimensional correlation NMR. The chemical shifts of the core cellulose that are similar to those of the $I_{β}$ allomorph are underlined.

<table>
<thead>
<tr>
<th>Organisms</th>
<th>Form</th>
<th>C1 (ppm)</th>
<th>C2 (ppm)</th>
<th>C3 (ppm)</th>
<th>C4 (ppm)</th>
<th>C5 (ppm)</th>
<th>C6 (ppm)</th>
<th>Sources</th>
</tr>
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<tr>
<td>Arabidopsis</td>
<td>Core</td>
<td>105.5/104.1</td>
<td>71.7</td>
<td>75.0</td>
<td>88.8</td>
<td>71.7</td>
<td>65.6</td>
<td>1.5 s PDSD</td>
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<tr>
<td>Brachypodium</td>
<td>Core</td>
<td>105.5/104.3</td>
<td>71.9</td>
<td>75.0</td>
<td>88.9</td>
<td>71.9</td>
<td>65.5</td>
<td>3.0 s PDSD</td>
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<tr>
<td>Cladophora</td>
<td>$I_{α}$</td>
<td>105.0</td>
<td>71.6</td>
<td>74.7</td>
<td>90.0</td>
<td>70.1</td>
<td>65.2</td>
<td>Kono et al., 2003</td>
</tr>
<tr>
<td>Tunicate</td>
<td>$I_{β}$</td>
<td>106.1</td>
<td>71.0</td>
<td>74.2</td>
<td>88.9</td>
<td>72.2</td>
<td>65.0</td>
<td>Kono et al., 2003</td>
</tr>
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</table>

Table 3. Surface to interior cellulose number ratios of various primary cell walls from $^{13}$C quantitative DP spectra and short-CP spectra.

<table>
<thead>
<tr>
<th>Sample</th>
<th>Preparation</th>
<th>Experiment</th>
<th>Peaks</th>
<th>Ratio</th>
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</thead>
<tbody>
<tr>
<td>Brachypodium</td>
<td>Iowa State, never-dried</td>
<td>Quantitative DP</td>
<td>s : i</td>
<td>1.4</td>
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<td></td>
<td></td>
<td>Short CP</td>
<td>s : i</td>
<td>1.2</td>
</tr>
<tr>
<td>Poa annua</td>
<td>U Kentucky, rehydrated</td>
<td>Quantitative DP</td>
<td>s : i</td>
<td>1.3</td>
</tr>
<tr>
<td>Arabidopsis, xxt1xxt2xxt5</td>
<td>Iowa State, rehydrated</td>
<td>Quantitative DP</td>
<td>s : i</td>
<td>1.2</td>
</tr>
<tr>
<td>Arabidopsis, WT, intact</td>
<td>Penn State, never-dried</td>
<td>Quantitative DP</td>
<td>(s+G) : i</td>
<td>1.8</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Short CP</td>
<td>(s+G) : i</td>
<td>1.5</td>
</tr>
<tr>
<td>Arabidopsis, digested wall</td>
<td>Penn State, never-dried</td>
<td>Quantitative DP</td>
<td>(s+G) : i</td>
<td>1.5</td>
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<tr>
<td>Arabidopsis, WT intact</td>
<td>U Kentucky, rehydrated</td>
<td>Quantitative DP</td>
<td>(s+G) : i</td>
<td>1.9</td>
</tr>
<tr>
<td>Arabidopsis, cesa1$^{a}$ges/cesa3$^{x}$rxr1-2</td>
<td>U Kentucky, rehydrated</td>
<td>Quantitative DP</td>
<td>(s+G) : i</td>
<td>2.5</td>
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</tbody>
</table>
Figure Legends

**Figure 1.** 2D $^{13}$C-$^{13}$C J-INADEQUATE spectra of never-dried *Arabidopsis* cell walls at room temperature, correlating double-quantum (DQ) and single-quantum (SQ) $^{13}$C chemical shifts. The spectra were measured at 400 MHz, 600 MHz and 900 MHz. Insets magnify the C1 region of the spectra to indicate the resolution enhancement by higher magnetic fields. The bottom row amplifies the C2-C4 region of arabinose, where high magnetic fields significant improve the resolution of multiple forms of arabinose.

**Figure 2.** 2D $^{13}$C-$^{13}$C PDSD spectra of plant cell walls measured with 1.5 s spin diffusion mixing. (a) Intact *Arabidopsis* cell wall at 20˚C. (b) HG-depleted *Arabidopsis* cell wall at -20˚C. (c) *Brachypodium* cell wall at -20˚C. Cellulose-pectin cross peaks are observed in both intact and HG-depleted *Arabidopsis* cell walls, and cellulose-GAX cross peaks are detected in the *Brachypodium* sample.

**Figure 3.** (a) 2D $^{13}$C-$^{13}$C PDSD spectrum of *Brachypodium* cell wall with a 3.0 s mixing. The spectrum was measured at 20˚C with a short $^1$H-$^{13}$C CP contact time of 35 μs to suppress the signals of mobile GAX. (b) Representative cross sections of interior cellulose (black) and surface cellulose (orange). The different intensity patterns indicate that $^{13}$C magnetization has not equilibrated between interior and surface cellulose. The difference spectra (purple), obtained after normalizing the two cross sections by the sC4 peak, correspond to core cellulose chains that are inaccessible to the surface. (c) Illustration of the cellulose microfibril structure, where interior cellulose consists of a surface-bound fraction and a core fraction. (d) The two types of interior cellulose chains have slightly different C6 chemical shifts.

**Figure 4.** 2D $^{13}$C-$^{13}$C PDSD spectrum of *Arabidopsis* cell walls with a 1.5 s mixing time. The spectrum was measured at -20˚C under 9 kHz MAS. (a) 2D spectrum. (b) Representative cellulose cross sections of interior and surface cellulose exhibit different intensity patterns. The difference spectra (purple) were obtained after normalizing the two spectra by the sC4 peak. The surface cellulose cross section has contribution from Ara and XyG backbone, but the difference spectra mainly show signals of interior cellulose. (c) C1 and C6 regions of the cellulose cross sections and the difference spectra. Core cellulose C1 shows two peaks at 105.5 ppm and 104.1 ppm, and core cellulose C6 (cC6) exhibits a 0.3-ppm downfield shift from the average interior cellulose C6 (iC6) and 0.6-ppm downfield shift from the surface-bound interior cellulose (bC6).

**Figure 5.** 1D quantitative $^{13}$C DP spectra of $^{13}$C-labeled primary cell walls at ambient temperature. All spectra were measured with recycle delays of 15 to 25 s, except for the xxt1xxt2xxt5 sample, which was measured with recycle delays of 10 s. (a) Spectra of grass cell walls with negligible amounts of XyG. Two grasses, *Brachypodium distachyon* (top) and *Poa annua* (bottom), were measured. The Ara C1 (AC1) and interior cellulose C4 (iC4) peaks are highlighted in green and red, respectively. The mixed peaks of surface cellulose C4 and Ara C2 and C4 are shaded in grey. The integrated intensities were used to calculate the surface : interior cellulose ratio (s : i). Grass has a small s : i ratio of 1.3 - 1.4, indicating at least 24 glucan chains (see **Figure 6**). (b) A triple mutant of *Arabidopsis thaliana* with negligible XyG. (c) Intact (top) and digested (bottom) *Arabidopsis* cell walls. (d) WT and CESA mutant of *Arabidopsis*. The integration regions are 111.8-107.2 ppm for AC1, 92.0-86.8 ppm for iC4 and 86.8-80.4 ppm for the mixed peak of sC4 and matrix polysaccharides. The boundary of the mixed peak changed to 81.0 ppm for the xxt1xxt2xxt5 mutant cell wall to avoid overlap with a strong pectin peak at 79.6 ppm.

**Figure 6.** Number of glucan chains in cellulose microfibrils as a function of the s : i ratio. The minimum number of glucan chains for s : i values of 1.1, 1.2, 1.3, 1.4 and 1.5 are 30, 29, 28, 24 and
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