

Copyright is owned by the Author of the thesis. Permission is given for a copy to be downloaded by an individual for the purpose of research and private study only. The thesis may not be reproduced elsewhere without the permission of the Author.

# **Development of Food-grade Microparticles from Lignin**



**A thesis presented in partial fulfilment of the requirements for the  
degree of**

**Doctor of Philosophy**

**in**

**Food Technology**

**at Massey University, Palmerston North, New Zealand**

**Hayley Elizabeth Stewart**

**2015**



## Abstract

An opportunity presented itself to develop a new food ingredient from the wood biopolymer lignin. Lignin is set to become a high-yielding and inexpensive by-product of lignocellulosic biorefining processes. The aromatic heteropolymer is water insoluble, relatively hydrophobic, non-digestible in its native form, and also considered to be non-toxic. Retention of these properties in isolated lignin may enable a microparticulate food ingredient, with particular use as a fat mimetic, to be developed.

Lignin was extracted from shrub willow using an organosolv pulping procedure. A reactor vessel suitable for pulping under organosolv conditions was obtained, modified and commissioned. Using central composite design, an ethanol concentration of 60% (v/v), extraction time of 132 minutes and extraction temperature of 195°C were found to extract lignin of the highest yield and purity. The total lignin content of lignin extracted from chipped, untreated willow under these conditions was  $96.53 \pm 0.10\%$  (w/w).

An anti-solvent precipitation technique was explored for the ability to produce lignin microparticles. An aqueous-organic solution of lignin was dispersed into a much larger volume of water, whereby leaching of organic solvent from the dispersed phase into the water resulted in progressive precipitation of the lignin solute, ultimately producing a suspension of solid microparticles. Temperature and addition of surfactant were investigated as a means of controlling particle properties. Increasing initial water temperature between 4 and 80°C produced increasingly large agglomerates of uniform sub-micron primary particles, increasingly fused into monolithic masses. Faster removal of ethanol, hydrophobic interactions or an increase in rate of diffusion limited coalescence may be responsible for the increased fusion at higher temperatures.

Incorporating the ionic surfactant sodium dodecyl sulfate (SDS) or cetyltrimethylammonium bromide (CTAB) in the water at temperatures of 20-60°C during anti-solvent precipitation produced spherical, monodisperse lignin particles in the range 0.1-0.2  $\mu\text{m}$ . However, concentrations of these surfactants greater than 1% (w/v) caused aggregation of primary particles, probably due to bridging between bilayers of surfactant at high concentration. The non-ionic surfactants Tween 20 and sucrose ester were unable to prevent particle fusion at the 60°C preparation temperature, indicating that the electrostatic repulsion between particles coated with SDS and CTAB is important to stability.

The ability to manipulate the size of SDS-stabilised particles using added salt and adjustment of pH was determined. The electrostatic mechanism of stabilisation by SDS was confirmed by the ability of added potassium chloride and calcium chloride to destabilise lignin particles. Measured particle size increased with the concentration of added salt, due to either greater fusion of lignin during particle formation or to aggregation of primary particles, depending on whether the salt was added before or after particle formation. Decreasing the pH of the suspension of particles to 1.5, which is lower than the pKa of SDS, caused primary particles to aggregate. For destabilisation with salt and pH, generally only the relative volumes of particles in two modal distributions ( $\sim 0.1\text{-}2\ \mu\text{m}$  and  $\sim 1\text{-}100\ \mu\text{m}$ ) appeared to change. This indicates that the ability to control particle size may be limited by aggregation kinetics. Adjustment and control of pH throughout particle formation may enable the preparation of particles in the intermediate size range, such as narrowly distributed  $1\text{-}10\ \mu\text{m}$  particles. The ability to prepare particles of various and controlled size is important for maximising industry applications.

To highlight a possible application of food-grade lignin microparticles, the performance of particles as Pickering stabilisers of oil-in-water emulsions was evaluated. A literature method of emulsion formation involving the redispersion of dried lignin particles and various pH-modification steps, and a method using lignin in the form of freshly prepared microparticle suspensions were explored. Both methods produced emulsions stable to coalescence for five months of chilled storage, although the pH-modification method produced slightly smaller ( $D[3,2]$  of  $2.7\text{-}11.9\ \mu\text{m}$ ) oil droplets than the fresh suspension method ( $D[3,2]$  of  $11.6\text{-}31.2\ \mu\text{m}$ ). It may be possible in the future to produce smaller emulsion droplets by using a higher energy emulsification technique and by utilising sub-micron lignin particles, rather than  $\sim 10\ \mu\text{m}$  particles.

Three main areas in which work could be carried out to move this project forward have been identified. These areas are:

1. Lignin characterisation – including molecular weight, chromatography to determine lignin fragment types and contact angle.
2. Control of particle size – including the investigation of more food-grade surfactants, ultrasonic mixing and controlled pH changes during anti-solvent precipitation.

3. Food applications – including further investigation into Pickering emulsions, foam stabilisation and fat mimetic applications.

## **Acknowledgements**

I would like to thank my supervisors, Matt Golding, Lara Matia-Merino, Clive Davies and Richard Archer for all their help in this project. Especially Lara and Matt for your expertise, practical advice, and for making me feel more at ease. Thanks Clive for going through things very thoroughly and always having a good story to tell. Thanks Richard for your many ideas, and also for pointing out so many split infinitives that I'll probably pick up on them for the rest of my life. I'll be sure to carefully avoid them from now on. See what I did there?

Many thanks to Byron McKillop for all his work on the extraction vessel and other pieces of equipment throughout the project – it could definitely not have gone ahead without your help. Similarly, to Garry Radford for always having the time to help and for coming up with a lot of ideas. In particular, thanks for staying late on a Friday night to help me peel the bark off willow sticks after helping me for two days already with it. Thanks to Warwick Johnson for your sense of humour and never making anything seem like a hassle. Thanks to Michelle Tamehana for your help with the Mastersizer and Zetasizer and for letting me know when there was beer that needed drinking. Thanks to Steve Glasgow for all your knowledge in the lab and helping with the willow chip processing. Thanks to Doug Hopcroft and Matthew Savoian for carrying out and helping me with SEM and confocal microscopy. Big thanks to Kirk Torr, Daniel van de Pas, Claire Armstrong and Sunita Jeram at Scion for sharing your lignin knowledge and carrying out lignin analyses for me. Many thanks to Ian McIvor from Plant and Food Research for sharing your extensive willow knowledge and for your support at my confirmation presentation. Thanks also to Mark Waterland for the discussions we had about lignin structure and for your advice. And thanks to all the other people who helped, including Kylie Foster, Catherine Hobbs, Robert Southward, Allan Hardacre, Yvonne Parkes, Christine Ramsay, Heather McClean and Ansley Te Hiwi.

I really appreciate the scholarship provided to me by the Riddet Institute and the Earle Food Research Fund to carry out my PhD. I would also like to thank Zonta Manawatu and the Graduate Women Manawatu Charitable Trust for awarding me a scholarship that enabled me to present my work at the Food and Biosystems Engineering Conference in Greece.

Cheers (more like chur) to my office mates for their encouragement and for putting up with some epic rants. Also, for getting involved in “flag of the day”, “rage of the day” and the (mandatory) office dance breaks. Special thanks to Sandra for helping me to practice for my presentations, and watching them enough to almost be my understudy. Thanks also to the other postgrads and 4<sup>th</sup> years who were always up for a yarn, especially Shakti – my honorary 5<sup>th</sup> supervisor.

Finally, I would like to acknowledge my family and friends for their support. A huge thanks to Mum and Dad for providing never-ending encouragement, and for your financial support in the last stages. Mum – thanks so much for answering all my phone calls even though I’m sure you had way better things to do than listen to my worries. And cheers to the rest of the family too. The good times up at the Lake kept me going for sure! Thanks to all my flatmates over the years, especially Darnelle and Jess for making sure that I still played hard. Thanks heaps to Rob, Erin, Melinda and Aaron for your support as well. We’ve got this!



# Table of Contents

<b>Chapter One. Introduction.....</b>	<b>23</b>
<b>Chapter Two. Background.....</b>	<b>28</b>
2.1. Lignin occurrence and function .....	28
2.2. Lignin distribution .....	30
2.3. Lignin synthesis and structure .....	31
2.3.1. Synthesis.....	31
2.3.2. Structure .....	34
2.4. Effects of lignin on nutrition and health .....	35
2.4.1. Effects of lignin in the diet .....	35
2.5. Applications of lignin in food.....	38
2.5.1. Scientific opinion on the use of calcium lignosulfonates in foods.....	38
2.5.2. FDA regulatory status .....	39
2.6. Lignin extraction.....	40
2.6.1. Traditional pulping methods .....	40
2.6.2. Recovery of lignin from ethanol biorefineries .....	40
2.7. Lignin characterisation .....	60
2.7.1. Lignin yield and purity .....	60
2.7.2. Lignin structure and functional groups .....	61
2.7.3. Molecular weight analysis.....	65
2.7.4. Lignin solubility .....	67
2.7.5. Differential scanning calorimetry.....	68
2.8. Microparticulation .....	70
2.8.1. Factors influencing microparticulation by liquid-liquid dispersion.....	77
2.9. Particle characterisation.....	91

2.10. Pickering emulsions .....	96
2.10.1. Types of emulsions .....	96
2.10.2. Types of instability for oil-in-water emulsions .....	97
2.10.3. Emulsifiers .....	97
2.10.4. Pickering emulsifiers .....	98
2.10.5. Examining the potential of particles to act as Pickering emulsifiers .....	99
2.10.6. Lignin as a Pickering stabiliser .....	104
<b>Chapter Three. Lignin Extraction Vessel .....</b>	<b>107</b>
3.1. Preliminary trials: Existing extraction vessel .....	107
3.2. Parr extraction vessel .....	107
3.2.1. Modifications to the Parr vessel .....	108
3.2.2. Testing the Parr vessel .....	109
3.3. Safe operating procedure .....	112
<b>Chapter Four. Lignin Extraction .....</b>	<b>113</b>
4.1. Materials and methods .....	113
4.1.1. Feedstock preparation .....	113
4.1.2. Extraction of lignin .....	116
4.1.3. Lignin analysis .....	118
4.1.4. Optimisation of lignin extraction conditions .....	119
4.2. Results .....	121
4.2.1. Screening .....	122
4.2.2. Optimisation by central composite design .....	123
4.2.3. Composition of lignin produced using optimised extraction conditions .....	127
4.3. Discussion .....	128
4.4. Conclusions and recommendations .....	129

<b>Chapter Five. Manufacture of Lignin Microparticles .....</b>	<b>130</b>
5.1. Introduction.....	130
5.2. Materials and methods .....	132
5.2.1. Materials.....	132
5.2.2. Methods.....	132
5.3. Results.....	136
5.3.1. Effect of temperature.....	136
5.3.2. Effect of sodium dodecyl sulfate.....	143
5.4. Discussion.....	146
5.4.1. Effect of temperature.....	146
5.4.2. Effect of SDS.....	148
5.5. Conclusions and recommendations .....	149
<b>Chapter Six. Control of Lignin Microparticle Size.....</b>	<b>150</b>
6.1. Introduction.....	150
6.2. Materials and methods .....	153
6.2.1. Materials.....	153
6.2.2. Methods.....	153
6.3. Results.....	157
6.3.1. Effect of SDS concentration on particle size.....	157
6.3.2. Effect of CTAB, Tween 20 and sucrose ester on lignin particle size .....	163
6.3.3. Controlled destabilisation of lignin microparticles as a tool for particle size manipulation.....	170
6.3.4. Control of hydrophobic interactions during particle formation .....	179
6.3.5. Effect of ultrasonic mixing on particle size.....	180
6.3.6. Re-dispersibility of lignin particles .....	181
6.4. Discussion.....	181

6.4.1. Effect of emulsifier type .....	182
6.4.2. Effect of emulsifier concentration .....	183
6.4.3. Effect of salt concentration.....	186
6.4.4. Effect of pH .....	187
6.4.5. Effect of urea .....	188
6.5. Conclusions and recommendations .....	189
<b>Chapter Seven. Lignin Microparticles as Pickering Stabilisers .....</b>	<b>191</b>
7.1. Introduction.....	191
7.2. Materials and methods .....	192
7.2.1. Materials .....	192
7.2.2. Methods .....	192
7.3. Results.....	196
7.3.1. Effect of lignin concentration .....	196
7.3.2. Effect of oil concentration .....	200
7.3.3. Comparison of the fresh suspension and pH-modification methods.....	214
7.4. Discussion.....	219
7.4.1. Emulsion appearance .....	219
7.4.2. Effect of lignin and oil concentration .....	220
7.4.3. Differences between the FS and pH-modification methods.....	221
7.4.4. Size of the lignin particles used for stabilisation.....	222
7.5. Conclusions and recommendations .....	224
<b>Chapter Eight. Overall Discussion and Conclusions .....</b>	<b>226</b>
8.1. Can food-grade lignin be extracted from wood? .....	226
8.2. Can the structure and functional properties of lignin be controlled?.....	228
8.3. Can the lignin microparticles act as Pickering stabilisers of emulsions? .....	231

8.4. Can lignin microparticles be used in other food applications?..... 233

8.5. Summary of recommendations ..... 234

## List of Tables

Table 1. Research questions and objectives .....	27
Table 2. Proportion of lignin in some lignocellulosic materials .....	28
Table 3. Summary of lignin produced using traditional pulping methods.....	42
Table 4. Influence of different cooking additives on Kappa number and total yield in ethanol-water pulping of <i>Eucalyptus globulus</i> (adapted from Gilarranz et al., 1998)....	56
Table 5. Working methods for FTIR analysis of lignin .....	63
Table 6. Working methods for NMR analysis of lignin used by different researchers ..	64
Table 7. Working methods for molecular weight analysis of lignin used by different researchers.....	66
Table 8. Glass transition temperatures of various lignins .....	69
Table 9. Working methods for DSC analyses of lignin used by different researchers ...	69
Table 10. Details of various microparticulation methods – how the method works, implications, examples of use and reference sources.....	71
Table 11. Applications, important characteristics and production methods of relevant microparticulate systems .....	75
Table 12. Details of characterisation techniques used for various microparticulate systems .....	92
Table 13. Names and proportions of the five wood chip fractions used for lignin extraction experiments .....	114
Table 14. Untreated <i>Salix purpurea</i> composition .....	115
Table 15. Oven-dry mass of each chip size fraction used in screening trial extraction experiments .....	116
Table 16. Experimental design matrix for optimisation of lignin extraction.....	121
Table 17. Details of secondary trials in the optimisation of lignin extraction conditions .....	121
Table 18. Influence of wood type, temperature, extraction time and ethanol concentration on lignin yield.....	122
Table 19. Influence of ethanol concentration and extraction time on lignin yield and purity .....	124
Table 20. Influence of extraction time on lignin yield and purity .....	126
Table 21. Composition of lignin extracted from untreated willow .....	127
Table 22. D[4,3] and D[3,2] values of lignin particles in suspensions prepared using initial water temperatures of 4-80°C .....	137
Table 23. Mass ratio of SDS to precipitated lignin at various SDS concentrations.....	158
Table 24. D[4,3] and D[3,2] values of lignin particles in suspensions stabilised by various concentrations of SDS and CTAB .....	164
Table 25. D[4,3], D[3,2] and specific surface area values of droplets in 10% (w/v) soybean oil emulsions containing 0.5-2.9% (w/v) lignin, prepared using the FS method.....	197

Table 26. D[4,3] and D[3,2] values of droplets in 10% (w/v) soybean oil emulsions containing 0.05-2% (w/v) lignin, prepared using the pH-modification method.....	199
Table 27. D[4,3] and D[3,2] values of droplets in emulsions prepared using the FS method with 1.39% (w/v) lignin and 3.8-28.6% (w/w) soybean oil.....	201
Table 28. D[4,3] and D[3,2] values of droplets in emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and 3.8-28.6% (w/w) oil.....	208
Table 29. D[4,3] and D[3,2] values of droplets in emulsions containing oil concentrations of 3.8-28.6% (w/w) and prepared using either the pH-modification (0.5% lignin) or FS (1.39% lignin) method.....	214
Table 30. Final pH of emulsions prepared using the pH-modification method and FS method with oil concentrations of 3.8-28.6% (w/w) .....	218

## List of Figures

Figure 1. Products of a lignocellulosic biorefinery .....	24
Figure 2. Cellulose strands surrounded by hemicellulose and lignin.....	29
Figure 3. The primary precursors of lignin: (a) <i>trans-p</i> -coumaryl, (b) <i>trans</i> -coniferyl, and (c) <i>trans</i> -sinapyl. ....	32
Figure 4. Representation of different linkages formed during enzymatic dehydrogenation between two <i>trans</i> -coniferyl units: (a) biphenyl carbon-carbon linkage, (b) alkyl-aryl carbon-carbon linkage, and (c) ether linkage.....	32
Figure 5. Common notation for denoting the different carbon atoms within each phenyl propane unit .....	33
Figure 6. Proposed structure of beech lignin. ....	35
Figure 7. Effect of ethanol concentration and extraction temperature on the yield of organosolv ethanol lignin.....	53
Figure 8. Effect of catalyst concentration and time on the yield of organosolv ethanol lignin. ....	55
Figure 9. FTIR spectra of commercial alkali lignin and lignin isolated from oil palm waste.....	62
Figure 10. Examples of methods used for the preparation of microspheres: (a) the “fed batch” method, (b) the flow through mixing system and (c) the “plug flow precipitation” method.....	80
Figure 11. SEM images of zein nanoparticles prepared from shearing a zein stock solution (1g of zein in 85% aqueous ethanol) into deionized water at a homogenisation speed of (A) 5000, (B) 10,000, and (C) 15,000 rpm.....	82
Figure 12. Effect concentration of ethanol in stock solutions on the average diameters of zein nanoparticles.....	86
Figure 13. Top: Positioning of hydrophilic (left), intermediate hydrophobicity (centre) and hydrophobic (right) particles at an air or oil/water interface, showing contact angles $< 90^\circ$ , equal to $90^\circ$ and $> 90^\circ$ , respectively. Bottom: The curvature of the interface depending on the particle contact angle; aqueous foams or o/w emulsions formed when $\theta < 90^\circ$ and aerosols or w/o emulsions formed when $\theta > 90^\circ$ .....	100
Figure 14. Major steps in the production of pH-responsive Pickering emulsions (styrene-in-water) prepared using lignin and an NaOH/HCl system (note: St = ten minutes of standing time).....	105
Figure 15. Schematic diagram (not to scale) of cross-section of modified reactor, with gasket, split ring assembly, compression ring and drop band excluded for clarity. ....	110
Figure 16. Final set-up of the Parr extraction vessel (quench water not shown).....	111
Figure 17. Surface plot for the effect of ethanol concentration and extraction time on lignin purity.....	125



Figure 18. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL water at temperatures of 4, 20, 40, 60 and 80°C.....	136
Figure 19. Photographs of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL water at temperatures of (from left to right) 4, 20, 40, 60 and 80°C.....	138
Figure 20. SEM images of the dried, crushed lignin microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water at temperatures of (A) 4°C, (B) 20°C, (C) 40°C, (D) 60°C and (E) 80°C. All images were taken at a magnification of 20,000X (bar scale = 3µm). ....	139
Figure 21. SEM image of the dried, crushed microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water at 4°C, stirring for 22 hours to remove ethanol and subsequently heating the particle suspension to 60°C. The image was taken at a magnification of 20,000X (bar scale = 3 µm). ....	140
Figure 22. FTIR spectra for dried, crushed lignin particles prepared by shearing 30 mL of lignin extraction liquor into 75 mL water at temperatures of 4, 20, 40 and 60°C....	140
Figure 23. DSC curve for dried, crushed lignin particles prepared by shearing 30 mL of lignin extraction liquor into 75 mL water at temperatures of 4, 20 and 60°C. ....	142
Figure 24. SEM images of dried, crushed lignin microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water at temperatures of (A) 45°C and (B) 50°C. The images were taken at a magnification of 20,000X (bar scale = 3 µm).....	142
Figure 25. SEM images of dried, crushed lignin microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water at 4°C, stirring for 22 hours to remove ethanol and subsequently heating the particle suspension to (A) 35.7°C and (B) 51.4°C. The images were taken at a magnification of 20,000X (bar scale = 3 µm).....	143
Figure 26. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL water containing 0.5% (w/v) SDS at temperatures of 4, 20, 40 and 60°C.....	144
Figure 27. SEM image of the dried, crushed lignin microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL water containing 0.5% (w/v) SDS at 60°C (bar scale = 3 µm). ....	145
Figure 28. Appearance of lignin extraction liquor before (left) and after (right) centrifugation at 98,922 x g. ....	145
Figure 29. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water (60°C) containing SDS concentrations of 0-1.5% (w/v).....	158
Figure 30. SEM images of microparticles prepared using an aqueous phase temperature of 60°C with (A) 0.0625% (w/v) SDS and (B) 1% (w/v) SDS (bar scale = 3 µm). ....	159
Figure 31. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of extraction liquor into 75 mL of water (60°C) containing an SDS concentration	

of 1% (w/v) that were subsequently treated with different periods of sonication time before laser light scattering measurements were made.....	160
Figure 32. Cryo-SEM image of liquid suspension of lignin microparticles prepared using 1% (w/v) SDS and water temperature of 60°C. ....	161
Figure 33. Cryo-SEM of liquid suspension of lignin microparticles prepared using 0.5% (w/v) SDS and water temperature of 60°C. ....	161
Figure 34. Particle size distributions of particle suspensions prepared by shearing 30 mL of extraction liquor into 75 mL of water (4°C) containing SDS concentrations of 0.125, 0.25 and 0.5% SDS (w/v).....	162
Figure 35. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL water (60°C) containing CTAB concentrations of 0-1.5% (w/v).....	163
Figure 36. Zeta potential of particles prepared using SDS concentrations of 0-1.5% (w/v). ....	165
Figure 37. Zeta potential of particles prepared using CTAB concentrations of 0-1.5% (w/v). ....	165
Figure 38. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL of water (60°C) containing 10-25% Tween 20.....	166
Figure 39. Particle size distributions of microparticle suspensions prepared by shearing 30 mL of lignin extraction liquor into 75 mL water (60°C) containing sucrose ester at concentrations of 0-2.5% (w/v).....	167
Figure 40. Size distributions of lignin microparticles prepared using various emulsifiers. ....	168
Figure 41. SEM images of dried, crushed lignin microparticles prepared by shearing 30 mL of lignin extraction liquor into 75 mL water containing (A) 0.5% (w/v) SDS, (B) 0.5% (w/v) CTAB, (C) 10% (w/v) Tween 20 and (D) 2.5% (w/v) sucrose ester at 60°C (bar scale = 3 µm). ....	169
Figure 42. Size distributions of lignin microparticles prepared using a water phase containing both 0.25% (w/v) SDS and 0-0.1 M KCl at 60°C.....	170
Figure 43. Size distributions of lignin microparticles prepared using a water phase containing both 0.5% (w/v) SDS and 0-0.1M KCl at 60°C.....	172
Figure 44. SEM images of lignin microparticles formed in the presence of 0.5% (w/v) SDS and (A) 0.05 M KCl, (B) 0.06 M KCl and (C) 0.1 M KCl, all at 60°C (bar scale = 3 µm). ....	172
Figure 45. Size distributions of lignin microparticles prepared using a water phase at 60°C containing 0.25% (w/v) SDS, with 0-0.1 M KCl added after the 22-hour stirring period of the lignin particle formation process. ....	173
Figure 46. SEM images of lignin microparticles prepared using a water phase containing 0.5% (w/v) SDS at 60°C where KCl has been added after the 22 hour stirring	

process of microparticle formation at concentrations of (A) 0.05 M and (B) 0.1 M KCl (bar scale = 3 $\mu\text{m}$ ). .....	174
Figure 47. Bar graph of zeta potential values measured for particle suspensions containing 0.25% (w/v) SDS and 0-0.1M KCl, which was added either before or after microparticle formation.....	175
Figure 48. Size distributions of lignin microparticles prepared using a water phase containing 0.5% (w/v) SDS where $\text{CaCl}_2$ has been added to the suspension after microparticle formation at concentrations of 5.1-20 mM. ....	176
Figure 49. Size distributions of lignin microparticles prepared using a water phase containing 0.5% (w/v) SDS, with pH adjusted to 3, 10 or 7.4 (unadjusted pH) and temperature adjusted to 60°C before mixing with the extraction liquor. ....	177
Figure 50. Size distributions of lignin microparticles prepared using a water phase containing 0.5% (w/v) SDS at 60°C, where the pH of the suspension has been adjusted after the 22 hour stirring process of microparticle formation to pH 1.5, 2, 7 or 10.....	178
Figure 51. SEM image of particles prepared using a water phase containing 0.5% SDS (w/v) at 60°C where the pH of the suspension has been adjusted to pH 1.5 after the 22 hour stirring process of microparticle formation (bar scale = 3 $\mu\text{m}$ ). ....	178
Figure 52. Size distributions of lignin microparticles prepared using water phases containing either water alone or varying concentrations of urea, at either 20 or 60°C.	179
Figure 53. SEM images of particles prepared using water phases containing (A) 6.6 M urea and (B) 8 M urea, at 60°C with no SDS present (bar scale = 3 $\mu\text{m}$ ). ....	180
Figure 54. Size distributions of lignin microparticles prepared using water phases containing either 0.5% SDS (w/v) alone or both 0.5% SDS (w/v) and 6.6 M urea, at temperatures of either 4 or 60°C. ....	181
Figure 55. Mechanism of flocculation of carbon nanotubes via surfactant molecules (Rastogi et al., 2008).....	185
Figure 56. 10% (w/v) soybean oil emulsions stabilised with 0.1, 0.5, 1.0, 1.4 and 2.9% (w/v) lignin using the FS method.....	196
Figure 57. Droplet size distributions in 10% (w/v) soybean oil emulsions containing 0.5-2.9% (w/v) lignin, prepared using the FS method. ....	197
Figure 58. 10% (w/v) soybean oil emulsions stabilised with 0.05, 0.1, 0.2, 0.5 and 2% (w/v) lignin using the pH-modification method. ....	198
Figure 59. Droplet size distributions in 10% (w/v) soybean oil emulsions containing 0.05-2% (w/v) lignin, prepared using the pH-modification method. ....	199
Figure 60. Droplet size distributions in emulsions prepared using the FS method with 1.39% (w/v) lignin and 3.8-28.6% oil (w/w). ....	200
Figure 61. Light microscopy images of droplets in emulsions prepared using the FS method with 1.39% (w/v) lignin and 3.8-28.6% (w/w) oil. ....	202
Figure 62. Confocal images of droplets in emulsions prepared using the FS method with 1.39% (w/v) lignin and either 13.8% or 28.6% (w/w) oil. Samples were stained with Nile Red before analysis (bar scale = 100 $\mu\text{m}$ ). ....	203

Figure 63. Photographs of emulsions prepared using the FS method with 1.39% (w/v) lignin and 3.8-28.6% (w/w) soybean oil. Photographs were taken two minutes after emulsion manufacture. ....	204
Figure 64. Photographs of emulsions prepared using the FS method with 1.39% (w/v) lignin and 3.8-28.6% (w/w) soybean oil. Photographs were taken ~45 minutes after emulsion manufacture. ....	205
Figure 65. Droplet size distributions in FS method emulsion containing 1.39% (w/v) lignin and 3.8% (w/w) oil on the day of manufacture and 2, 4, 8, 12 and 20 weeks after manufacture. ....	206
Figure 66. Droplet size distributions in FS method emulsion containing 1.39% (w/v) lignin and 28.6% (w/w) oil on the day of manufacture and 2, 4, 8, 12 and 20 weeks after manufacture. ....	206
Figure 67. Droplet size distributions in emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and 3.8-28.6% (w/w) oil. ....	207
Figure 68. Light microscopy images of droplets in emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and 3.8-28.6% (w/w) oil. ....	209
Figure 69. Confocal microscopy images of droplets in emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and either 13.8% or 28.6% (w/w) oil. Samples were stained with Nile Red before analysis (bar scale = 100 $\mu\text{m}$ ). ....	210
Figure 70. Photographs of emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and 3.8-28.6% (w/w) soybean oil. Photographs were taken two minutes after emulsion manufacture. ....	211
Figure 71. Photographs of emulsions prepared using the pH-modification method with 0.5% (w/v) lignin and 3.8-28.6% (w/w) oil. Photographs were taken ~45 minutes after emulsion manufacture. ....	212
Figure 72. Droplet size distributions in pH-method emulsion prepared using 0.5% (w/v) lignin and 3.8% (w/w) oil on the day of manufacture and 2, 4, 8, 12 and 20 weeks after manufacture. ....	213
Figure 73. Droplet size distributions in pH-method emulsion prepared using 0.5% (w/v) lignin and 26.8% (w/w) oil on the day of manufacture and 2, 4, 8, 12 and 20 weeks after manufacture. ....	213
Figure 74. Size distributions of lignin particles in the water phase used for the emulsification process in the pH-modification and FS methods. ....	215
Figure 75. SEM images showing lignin in water immediately prior to the addition of oil to form emulsions. The samples were (A) dried from the lignin dispersion at pH 3 in the pH-modification method (dispersion dried on glass slide for SEM analysis) and (B) dried from the lignin microparticle suspension (suspension centrifuged and the pellet dried and crushed for SEM analysis) (bar scale = 3 $\mu\text{m}$ ). ....	216
Figure 76. Light microscopy images of lignin microparticle dispersions before mixing with oil in (A) the FS method and (B) the pH-modification method (bar scale = 500 $\mu\text{m}$ ). ....	217

Figure 77. Zeta potential of 3.8 and 28.6% oil emulsions prepared using the pH-  
modification method (with 0.5% lignin) and the FS method (with 1.39% lignin). ..... 218  
Figure 78. Major steps and outcomes of project ..... 227

## List of Equations

Equation 1 .....	77
Equation 2 .....	89
Equation 3 .....	100
Equation 4 .....	103
Equation 5 .....	134
Equation 6 .....	134

**List of Appendices**

Appendix A ..... 256  
Appendix B ..... 260  
Appendix C ..... 264  
Appendix D ..... 269  
Appendix E ..... 273  
Appendix F ..... 276  
Appendix G ..... 283  
Appendix H ..... 288  
Appendix I ..... 306