



University of HUDDERSFIELD

University of Huddersfield Repository

Charles, C.J., Rout, S.P., Patel, K.A., Akbar, S., Laws, Andrew P., Jackson, B.R., Boxall, S.A. and Humphreys, Paul

Floc formation reduces the pH stress experienced by microorganisms living in alkaline environments

Original Citation

Charles, C.J., Rout, S.P., Patel, K.A., Akbar, S., Laws, Andrew P., Jackson, B.R., Boxall, S.A. and Humphreys, Paul (2017) Floc formation reduces the pH stress experienced by microorganisms living in alkaline environments. *Applied and Environmental Microbiology*, 83 (6). ISSN 0099-2240 (In Press)

This version is available at <http://eprints.hud.ac.uk/31009/>

The University Repository is a digital collection of the research output of the University, available on Open Access. Copyright and Moral Rights for the items on this site are retained by the individual author and/or other copyright owners. Users may access full items free of charge; copies of full text items generally can be reproduced, displayed or performed and given to third parties in any format or medium for personal research or study, educational or not-for-profit purposes without prior permission or charge, provided:

- The authors, title and full bibliographic details is credited in any copy;
- A hyperlink and/or URL is included for the original metadata page; and
- The content is not changed in any way.

For more information, including our policy and submission procedure, please contact the Repository Team at: E.mailbox@hud.ac.uk.

<http://eprints.hud.ac.uk/>

1 **Floc formation reduces the pH stress experienced by microorganisms living in**
2 **alkaline environments.**

3 C. J. Charles¹, S. P. Rout¹, K. A. Patel¹, S. Akbar¹, A. P. Laws², B. R. Jackson³, S. A.
4 Boxall³ and P. N. Humphreys¹.

5 ¹ Department of Biological Sciences, School of Applied Sciences, University of
6 Huddersfield, Queensgate, Huddersfield HD1 3DH, UK.

7 ² Department of Chemical Sciences, School of Applied Sciences, University of
8 Huddersfield, Queensgate, Huddersfield HD1 3DH, UK

9 ³Bio-imaging Facility, School of Molecular and Cellular Biology, Faculty of Biological
10 Sciences, University of Leeds, Leeds, LS2 9JT, UK.

11 **Corresponding Author:** Paul Humphreys, Department of Biological Sciences, School
12 of Applied Sciences, University of Huddersfield, Queensgate, Huddersfield HD1 3DH,
13 UK. Tel: (+44)1484 472771, Email: p.n.humphreys@hud.ac.uk

14 **Running title:** Microbial floc formation is a key alkaline survival strategy

15 **Abstract**

16 The survival of microorganisms within a cementitious geological disposal facility for
17 radioactive wastes is heavily dependent on their ability to survive the calcium
18 dominated, hyper-alkaline conditions resulting from the dissolution of the cementitious
19 materials. The present study shows that the formation of flocs, composed of a complex
20 mixture of extracellular polymeric substances (EPS), provides protection against
21 alkaline pH values up to pH 13.0. The flocs were dominated by *Alishewanella* and
22 *Dietzia sp*, producing a mannose rich carbohydrate fraction incorporating extracellular
23 DNA, resulting in Ca²⁺ sequestration. EPS provided a ~10 µm thick layer around the
24 cells within the centre of the flocs, which were capable of growth at pH 11.0 and 11.5,
25 maintaining internal pH values of pH 10.4 and 10.7 respectively. Survival was
26 observed at pH 12.0, where an internal floc pH of 11.6 was observed alongside a
27 reduced associated biomass. Limited floc survival (<2 weeks) was observed at pH
28 13.0. This study demonstrates that flocs are able to maintain a lower internal pH in
29 response to the hyperalkaline conditions expected to occur within a cementitious,
30 geological disposal facility for radioactive wastes and indicates that floc communities
31 within such a facility would be capable of survival up to a pH of 12.0.

32 **Importance**

33 The role of extracellular polymeric substances (EPS) in the survival of microorganisms
34 within hyperalkaline conditions is poorly understood. Here we present data for the
35 taxonomy, morphology and chemical characteristics of an EPS based microbial floc,
36 formed by a consortia isolated from an anthropogenic hyperalkaline site. Short term
37 (<2 weeks) survival of the flocs at pH 13 was observed with indefinite survival
38 observed at pH 12.0. Micro pH electrodes (ø10 µm) were utilised to demonstrate that

39 floccs were able to maintain a lower internal pH in response to hyperalkaline conditions
40 (pH 11.0, 11.5 and 12.0), demonstrating for the first time that floc formation and EPS
41 production is a survival strategy under hyperalkaline conditions. The results indicate
42 how microbial communities may survive and propagate within the hyperalkaline
43 environment expected to prevail in a cementitious geological disposal facility for
44 radioactive wastes, they are also relevant to the wider extremophile community.

45 **Introduction**

46 One of the concepts for the disposal of the UK's intermediate level radioactive waste
47 (ILW) inventory is a geological disposal facility (GDF) employing a cementitious backfill
48 (1, 2). It is anticipated that post closure, groundwater ingress into such a facility will
49 result in a saturated, anaerobic, calcium rich, hyper-alkaline environment that will aid
50 radionuclide retention through sorption and the formation of insoluble complexes (3,
51 4). These conditions will also result in the alkaline hydrolysis of the cellulose
52 component of the ILW producing a range of small molecular weight organic
53 compounds collectively known as cellulose degradation products (CDP) (5). The
54 biodegradation of CDP has received considerable attention in recent years with a
55 number of authors reporting alkaliphilic degradation under a wide range of growth
56 conditions (6, 7) including alkaliphilic methanogenic consortia operating at pH 11.0 (8).
57 In some cases these alkaliphilic communities have been shown to form floccs where
58 the bacteria are encased in a matrix of extracellular polymeric substances (EPS) (9).
59 The ability of freshwater and marine microorganisms to aggregate into a sustainable
60 microenvironment is documented (10, 11), however the added selective pressure of
61 alkaline pH is not commonly observed in the natural world.

62 EPS such as carbohydrates, proteins, lipids and extracellular DNA (eDNA) are
63 ubiquitous components of biofilm matrices (12). In addition to EPS; inorganic materials
64 (minerals) may also be incorporated to provide structural support where physical
65 stresses may impact on survival (13). Biofilm formation is known to enhance survival
66 against a range of environmental stresses such as pH shifts (14), with some biofilm
67 communities creating more favourable growth conditions through the secretion of
68 specific EPS components (15, 16). In these cases biofilm formation allows microbial
69 propagation in extreme environments (16, 17).

70 In the case of hyper-alkaline environments the primary environmental stress is the
71 extreme pH and there are examples in the literature of flocs and biofilms attenuating
72 the ambient pH. Aggregates of *Bacillus laevolacticus* modulated their internal pH by
73 between 0.4 – 2.0 pH units (18). A difference of 1 pH unit in methanogenic aggregates
74 from an up flow anaerobic sludge blanket have also been observed (19). Large pH
75 differences have been recorded in *Pseudomonas* biofilms; where a difference of 1.4
76 pH units was measured across distances of 50 μ m (20). Further pH shifts have been
77 seen across a variety of distances in a range of biofilms grown under different
78 conditions (21-23). However, in all these cases, the pH shifts are associated with near
79 neutral conditions and as such, the logarithmic nature of the pH scale means that the
80 modulation of internal pH in response to alkaline conditions represents a significantly
81 more difficult challenge to the microorganisms concerned.

82 Information regarding the ability of flocs to attenuate pH within alkaline environments
83 is currently absent from the literature. Consequently, the aim of this study was to
84 determine the ability of alkaliphilic flocs (9) to attenuate environmental pH values
85 relevant to a cementitious GDF. The isolated flocs were also characterised in terms of
86 microbial community structure, morphology and compositional aspects of the EPS

87 produced. The study sought to determine what degree floc formation is a survival
88 strategy for alkaliphilic microorganisms; and if flocs provide a potential dispersion
89 vehicle for alkaliphilic microorganisms within hyper-alkaline environments such as a
90 cementitious GDF.

91 **Materials and Methods**

92 **Community composition and flocculate concentration**

93 A previously described flocculate producing microcosm operating at pH 11.0 was sub-
94 sampled and maintained as described previously (9). Briefly, the microcosm was
95 inoculated with material retrieved from an area inundated with alkaline leachate at the
96 lime kiln waste site Harpur Hill, Derbyshire, UK. The microcosm was completely mixed,
97 incubated at 25 °C and fed alkaline cellulose degradation products every two weeks
98 on a 10% waste/feed cycle. Microcosm fluid (25 mL) was centrifuged at 5,000 x *g* for
99 10 minutes with the resulting pellet re-suspended in 25mL of phosphate buffer as
100 described by Hurt et al. (24) Genomic DNA was extracted for analysis using the
101 methods of Griffiths et al (25). The V4 region of the 16S rRNA gene was amplified
102 using primers 519F (5'CAGCMGCCGCGGTAA'3) and 785R
103 (5'TACNVGGGTATCTAATCC'3) for both bacteria and archaea (26, 27) with the
104 following overhangs 5' TCGTCGGCAGCGTCAGATGTGTATAAGAGACAG'3 and 5'
105 GTCTCGTGGGCTCGGAGATGTGTATAAGAGACAG'3, respectively. PCR reaction
106 mixture contained 20 ng of purified DNA solution, 0.5µL of each primer (20 pmol µL⁻¹
107 concentration), and 25 µL of MyTaq HS red mix (BIOLINE, UK) made up to 50 µL
108 volume with PCR grade water. The reaction mixture was then incubated at 94 °C for
109 1 minute, and then cycled 35 times through three steps: denaturing (94 °C, 15 s),
110 annealing (60 °C, 15 s), primer extension (72 °C, 10 s). This was followed by a final

111 extension step of 72 °C for 5 minutes. 16S rRNA gene microbial community analysis
112 was carried out via a MiSeq platform (Illumina, USA) at 250 bp paired ends with
113 chimera detection and removal performed via the UNCHIME algorithm in the Mothur
114 suite (28) (Chunlab, South Korea). Assignment of OTU's was performed using a CD-
115 HIT clustering method with a 95 % cut-off value with taxonomic assignment performed
116 against the EZtaxon database (29) .

117 Floc concentration was determined using a Guava easyCyte™ flow cytometer
118 (Millipore, US) with flocs stained using FITc (Fisher, UK) in accordance with methods
119 outlined in Chen, et al. (30). Floc size distribution was analysed using a Mastersizer
120 2000 (Malvern, UK) with a dispersant refractive index of 1.330 and a particle refractive
121 index of 1.572, with flocs isolated by centrifugation and re-suspended in ultrapure
122 water before analysis.

123 **Morphology**

124 The overall morphology of the flocs was investigated using a Quanta FEG 250
125 scanning electron microscope (SEM) and energy-dispersive X-ray spectroscopy
126 (EDX) used for analysis of sites of interest. Samples were fixed overnight in 4 %
127 paraformaldehyde (Fisher, UK) then dehydrated using a serial ethanol dilution of 25,
128 50, 75 and 100 % for 2 min per step. Samples were then dried onto a metal disc and
129 sputter coated via a gold palladium plasma (CA7625 Polaron, Quorum Technologies
130 Ltd, UK). The structural morphology of the flocculates was investigated via confocal
131 laser scanning microscopy (CLSM) at the Bio imaging centre of Leeds University using
132 a Zeiss LSM880 inverted confocal microscope with image analysis performed using
133 Zen 2.1 (Zeiss Microscopy). Flocs were fixed in 4% Paraformaldehyde (Fisher, UK)
134 overnight and then stained using the following compounds in accordance with
135 methods outlined in Chen et al. (30): Calcofluor white for the visualisation of β -1,4 and

136 β -1,3 polysaccharides (Sigma, UK), Nile red (Fisher, UK) for lipids and hydrophobic
137 sites, concanavalin A, tetramethylrhodamine conjugate (Fisher, UK) for α -
138 mannopyranosyl, α -glucopyranosyl sugars, FITc (Fisher, UK) for protein and Syto 63
139 (Fisher, UK) for total cells and extracellular DNA. Fluorescence in situ hybridisation
140 was carried out as per the methods of Ainsworth et al (31), using previously described
141 probes for the identification of total bacteria (32), Firmicutes (33), γ -Proteobacteria
142 (34) and Actinobacteria (35).

143 **EPS extraction, Purification and Characterisation**

144 Crude EPS was extracted from the flocs using a multiple extraction method outlined
145 by Ras et al. (36), which extracts different components of EPS based upon the
146 disruption of different chemical bonds. ATP was measured at each step to ensure the
147 EPS extraction was not causing the lysis of cells and thus contaminating the extracted
148 EPS. This was performed using a 3M Clean-Trace Biomass Detection Kit and
149 Luminometer (3M, UK) employing a modified method described previously by Charles
150 et al. (9); with CFU mL⁻¹ calculated using a standard curve of Escherichia coli K12
151 concentrations (data not shown). The crude EPS extracts were then measured for
152 carbohydrate content via the phenol sulphuric acid method (37), protein content via
153 the Bradford assay (38) lipid content via the methods of Bligh and Dyer (39) and the
154 DNA content via a Genova-nano spectrophotometer (Jenway, Bibby Scientific, UK).

155 In order to purify the extracted EPS, dialysis was carried out against ultrapure water
156 for 72 hours with the water changed every 24 hours. The protein and carbohydrate
157 fractions were then isolated from the dialysed EPS. Protein was precipitated via
158 treatment with trichloroacetic acid and the carbohydrate fraction precipitated via
159 ethanol treatment as outlined in Marshall, et al. (40). Monomer composition was
160 determined by TFA hydrolysis followed by anion exchange chromatography as

161 described previously (41) with the exception that sample was eluted with aqueous 0.01
162 M NaOH for 20 minutes followed by 83 % 10 mM NaOH: 17 % 150 mM NaOH:1 M
163 sodium acetate for 25 minutes for simultaneous determination of monomers and
164 uronic acids. Dry weight and inorganic content of the flocs were determined by
165 methods outlined in BS ISO 6496:1999 (42).

166 **EPS calcium binding assay**

167 Aqueous suspensions of EPS:Ca²⁺ (as CaCl₂) were prepared to concentrations of 0.1,
168 0.25, 0.5 and 1 g. g⁻¹ dry weight and incubated at room temperature for 15 minutes.
169 The calcium content of the samples was then quantified using a Metrohm 850
170 Professional IC (Metrohm, Cheshire, UK) with pulsed amperometric detection,
171 employing a Metrohm C4 Column (250 mm x 4.6 mm) with a mobile phase of 4.6 mM
172 phosphoric acid. All reactions were carried out under nitrogen to avoid interference
173 from atmospheric carbon dioxide and EPS bound calcium determined via comparison
174 with EPS-free incubations. Total bound calcium was determined using atomic
175 absorbance spectroscopy (Agilent 200 series AA, Agilent, UK) following digestion of
176 EPS (1 mg) in 10 ml of 0.1 % KCl: 1 % HNO₃.

177 **Floc profiling**

178 pH profiles of the flocs were undertaken with a micromanipulator and stand (Unisense,
179 Denmark) using a 10 µm diameter pH electrode with an external reference (Unisense,
180 Denmark) connected to a single channel pH/redox meter supplied by the probe
181 manufacturer (Unisense, Denmark). The probe was calibrated against pH 4.0, pH 7.0
182 and pH 10.0 standards (Fisher, UK) and tested against pH 11.0 and pH 12.0 solutions
183 made using NAOH. Profiles were taken through the flocs at pH 11.0, 11.5 and 12.0.
184 In order to generate the pH profile of the flocs, microcosm fluid was incubated for 1
185 hour at the desired pH and then injected into agar cubes of the same pH to provide

186 support for the profiling. Control profiles were conducted through agarose at pH 11.0,
187 11.5 and 12.0. In order to investigate floc stability under a range of pH values zeta
188 potential of the flocs was measured using a Zetasizer nano (Malvern, UK) with the
189 zeta potential calculated using Smoluchowski's equation. Flocs were isolated by
190 centrifugation and re-suspended in ultrapure water of the desired pH (values between
191 pH 7.0 and pH 12.0) before analysis.

192 **Floc survival**

193 In order to investigate the ability of the floc communities to survive prolonged exposure
194 to hyperalkaline conditions small scale microcosms of approximately 10^4 cells per ml
195 were formed using 100 ml Wheaton bottles at pH values 11.0, 11.5, 12.0 and 13.0 in
196 duplicate. The microcosms were composed of mineral media (43) and CDP to match
197 the composition of the original microcosm reported in Charles et al. (9). CDP was
198 produced by methods outlined previously by Rout et al.(44) and pH was measured on
199 a weekly basis using a portable handheld probe and meter (Mettler Toledo, UK), with
200 pH adjusted accordingly using sodium hydroxide or hydrochloric acid. The head space
201 of the microcosms was kept under nitrogen to ensure anaerobic conditions. The
202 microcosms were sampled for ATP concentration on a weekly basis for three weeks
203 using a 3M Clean-Trace Biomass Detection Kit and Luminometer (3M, UK) as
204 previously described. Abiotic controls were established for each pH value within the
205 pH range and were sampled as per the above.

206 **Accession number**

207 16S rRNA gene sequence data was uploaded to the NCBI sequence read archive
208 under the accession number SRP082489

209 **Results**

210 **Floc Characterisation**

211 Sequencing of the microbial 16S rRNA gene returned 18854 reads via Illumina MiSeq
212 technology. The microcosm community was dominated by sequence reads associated
213 with the Phyla Proteobacteria (39.49 %), Actinobacteria (32.93 %), Firmicutes (24.85
214 %) and Bacteroidetes (2.34 %) making up 99.6 % of the library (Fig. 1). The
215 Proteobacteria and Actinobacteria were each dominated by sequence reads
216 associated with *Alishewanella* (31.81 %) and *Dietzia* (26.74 %) respectively. Reads
217 associated with *Hydrogenophaga* and *Silanimonas* were also detected within the
218 Proteobacteria group, where *Leucobacter* and *Corynebacterium* represented >1 % of
219 the total reads within the Actinobacteria. The Firmicutes detected were not dominated
220 by a particular species *per se*, where *Anaerobranca* (4.55 %), *GQ356941_g* (4.45 %),
221 *Tissierella* (2.67 %), *Natronobacillus* (2.55 %), *Bacillus* (2.04 %) and *Bacillus_g26*
222 (1.68 %) were all represented within the sequence reads. The Bacteroidetes were
223 almost entirely composed of the sequence reads associated with the genus
224 *Aquiflexum* (2.16 % of the total reads).

225 Flow cytometry indicated that the floc concentration was 2.0×10^5 flocs/ml in the pH
226 11.0 microcosm with 54.4 % of the flocs being >10 μm in diameter reaching a
227 maximum size of 240 μm (Fig. S1). SEM investigations (Fig. S2) revealed individual
228 flocs to be clusters of cells, polymeric substance and crystalline precipitates. These
229 precipitates were composed elementally of calcium, carbon and oxygen indicating
230 calcium carbonate precipitation (Fig. S2). Confocal laser scanning microscopy (CLSM)
231 revealed the flocs to be composed of a complex mixture of proteins, carbohydrates,
232 lipids, eDNA and cells (Fig. 2). The most basal layer of the floc was composed primarily
233 of lipids and β -1,4 and β -1,3 polysaccharides, where the most outer layers of the floc

234 were composed of concentrated areas of proteins, these regions were surrounded by
235 α -mannopyranosyl and α -glucopyranosyl sugars which were also closely associated
236 with eDNA. These regions of pyranosyl sugars and eDNA surrounded the crystalline
237 precipitates observed under SEM. Bacterial cells were in the centre of the floc, with
238 $\sim 10 \mu\text{m}$ of EPS material surrounding these cells (Fig. S3). FISH/CLSM probing (Fig.
239 3) showed that the Actinobacteria (red) and γ -Proteobacteria (blue) were clustered
240 together, with Firmicutes (green) being situated amongst the periphery of these
241 clusters within the centre of the floc. The flocs ranged from 50 – 250 μm in diameter
242 and most featured a central well defined mass with looser less formed sections
243 attached. The denser areas of the flocs showed higher numbers of cells and more
244 concentrated areas of all EPS components.

245 The initial extraction of the floc associated EPS using sonication and ethanol
246 precipitation removed a primary, lipid rich EPS, consisting of 8.4 mg/g VS of lipids,
247 carbohydrates (3.2 mg/g VS) and low levels (<1 mg/g VS) of protein and eDNA (Fig.
248 S4). EDTA extraction yielded a significantly greater mass of volatile solids, with the
249 EPS extracted being carbohydrate rich (38.2 mg/g VS); again, eDNA and proteins
250 were also part of the EPS structure, with lipids being the least dominant component
251 following EDTA extraction. The extraction process was able to extract a combined
252 41.4 ± 3.7 mg/g VS carbohydrate, 5.0 ± 0.7 mg/g VS of eDNA, 9.7 ± 1.0 mg/g VS and
253 4.7 ± 0.2 mg/g VS protein with a dry weight content of the microcosm of 15.5 g/L of
254 which 44.4 % was volatile solids and 55.5 % was inorganic ash. Monomer analysis of
255 the carbohydrate fraction of the extracted EPS (Table 1) showed half the monomers
256 to be composed of mannose, with ribose and ribitol making up a further 20 %. A varied
257 range of sugars were identified in smaller amounts with uronic acids also present but
258 only making up a small proportion (4.3 %) of the total monomer composition.

259 The calcium content of the liquid component of the floc microcosm was 1.6 g/L which
260 was lower than that of the abiotic comparison which measured 4.3 g/L. Analysis of the
261 dialysed EPS via AA spectroscopy revealed 0.203 mg of calcium per mg of EPS.
262 Investigation into the binding capacity of dialysed EPS indicated that the EPS was able
263 to bind a further 0.173 mg/mg EPS (Fig. S5) giving a total binding capacity of 0.376
264 mg/mg EPS.

265 **Floc profiling and response to alkaline conditions.**

266 Analysis of the zeta potential of the flocs under a range of pH values showed
267 consistent stable potentials of -20 to -30 mV when transferred to solutions of pH 7-9
268 and 10-12 (Fig. 4A). When subjected to a pH of 10 the zeta potential shifted to $2.65 \pm$
269 0.36 mV. Following exposure of the flocs to elevated pH values representative of a
270 GDF, flocs demonstrated the ability to grow at pH 11.0 with cell numbers increasing
271 from a concentration of 2.5×10^4 CFU/ml to 1.8×10^6 CFU/ml (Fig. 4B). When subjected
272 to a pH of 11.5, the flocs showed only a small increase in cell concentration from 2.5
273 $\times 10^4$ CFU/ml to 4.8×10^4 CFU/ml. Cells within the floc were capable of survival when
274 sub cultured to pH 12.0, however the concentration of cells fell sharply from 7.7×10^3
275 CFU/ml to 4.1×10^2 CFU/ml within the first two weeks and then remained stable up to
276 the end of week 3. At pH 13 the flocs were able to maintain detectable cell
277 concentrations for two weeks, after which cells could not be detected. No increase in
278 ATP values were reported from control microcosms (data not shown).

279 The pH profiling was carried out on a sub sample of each of the surviving flocs (Fig.
280 4C) exposed to pH 11.0, 11.5 and 12.0. The interior of the flocs in all cases had a
281 lower pH value than the exterior pH with minimum pH values of 10.4, 10.7 and 11.6 at
282 pH 11.0, 11.5 and 12.0 indicating pH shifts found within the flocs were 0.6, 0.8 and 0.4
283 pH points at external pH values of 11.0, 11.5 and 12.0, respectively. These pH shifts

284 occurred over a short distances within the flocs with the largest pH shifts occurring
285 over a distance of 35µm within the flocs at each pH value tested. Control profiles
286 showed no change in pH values along profiles of similar lengths (data not shown).

287 **Discussion**

288 The work presented here provides the first comprehensive description of an alkaliphilic
289 floc based microbial community isolated from a calcium dominated, anthropogenic
290 hyperalkaline environment and demonstrates how adopting a floc based life style
291 protects the microbial community from the ambient pH. This further emphasises the
292 fact that it is the microenvironments which microorganisms create rather than the bulk
293 environmental chemistry that determine microbial success in the environment (10, 11).
294 The flocs isolated from this community were dominated by bacteria from the genera
295 *Alishewanella* and *Dietzia*. *Alishewanella* has been previously reported to form and
296 maintain flocs due to their ability to form biofilm and pellicles (45). Bacteria from the
297 genus *Dietzia* have been reported in a range of hyperalkaline areas and possess the
298 ability to degrade a range of carbohydrates and pollutants in planktonic or biofilm form
299 (46, 47). The large proportion of both *Alishewanella* and *Dietzia* within the community
300 suggests they play a key role in the maintenance of the floc structure (48). Within the
301 EPS environment, these taxa were closely associated with each other, suggesting a
302 synergistic relationship which enhanced survival at these extremes of pH. A number
303 of the Firmicutes detected have been previously associated with alkaline conditions,
304 the diverse metabolic capabilities of these organism would contribute to the overall
305 metabolism capabilities of these flocs (49-51).

306 The carbohydrate fraction of the EPS was mannose rich, previous studies suggest
307 that mannose rich biopolymers are directly involved in the sequestration of calcium

308 species (52), the results obtained within the calcium binding assay supports this
309 conclusion. The eDNA component of the EPS although acidic in nature due to the
310 linkage of nucleotides by the 3' - 5' phosphodiester bonds, most likely plays a role in
311 calcium sequestering due to its thermodynamically favourable interaction with calcium
312 ions (53). Calcium has been shown to promote bioflocculation (54), as evidenced by
313 the production of flocs up to 240µm. Here, the sequestration of calcium appears to
314 play two key roles; structural support and the buffering of pH. The interactions with
315 eDNA and formation of carbonates provides structural support to the floc where SEM
316 investigations clearly showed that EPS was bound to calcium carbonate precipitates
317 and previous research has shown that eDNA maintains *Alishewanella* associated
318 biofilm structure (9).

319 CLSM imaging of the flocs suggested that the basal layer of the aggregate provides a
320 hydrophobic core to the aggregate. This increase in hydrophobicity would reduce the
321 transport of hydroxide ions into the centre of the flocs resulting in a lower core pH.
322 Acidic phospholipids are also associated with alkaliphilic bacterial membranes and
323 may also buffer against the external pH (55). Proteins coated the carbohydrate
324 fractions of the EPS, where the production of extracellular proteins has been
325 implicated with the promotion of flocculation (56), however these proteins may also
326 buffer the local environment through their acidic nature as previously observed in
327 alkaliphilic bacteria (57).

328 In their cultured state, the flocs had an internal pH of 10.4 in an external environment
329 of pH11.0. Following subculture to a fresh media at this pH, increase in biomass and
330 further floc production was observed. Zeta potential measurements indicated that floc
331 formation was favoured at pH values close to the internal pH of the flocs (pH 10-11).
332 Sub culturing at pH values >pH 10.0 resulted in decreased viability of the floc

333 community, with a complete cessation of growth at pH 13.0. Survival of the flocs was
334 observed at pH 11.5, where energy associated with the generation of biomass is likely
335 to have been diverted into the maintenance of the internal floc pH to \approx pH 10.7. The
336 subculturing of flocs at pH 12.0 resulted in the loss of biomass, whilst the flocs
337 remained stable. The floc internal pH was now pH 11.6, suggesting the survival of only
338 the most alkaliphilic microorganisms, with much of the energetic process used to
339 maintain the internal floc pH. Although the shift in pH may only have been 0.4 – 0.8
340 units, at these extremes of pH the differential in hydroxyl ion concentration between
341 the internal and external floc surfaces was substantial (between 0.25 to 4.0 mM).

342 The floc community described within this study was grown using the major carbon
343 source expected within a cementitious GDF for ILW and is the first to be evaluated for
344 its survival and propagation within the calcium dominated hyperalkaline conditions
345 expected within such a disposal concept. The ability of flocs to survive at pH 12.0 and
346 persist short term at pH 13.0 suggest that microbial communities will require regions
347 of lower pH (<pH 12.0) than that anticipated to dominate a cementitious GDF (pH 12.5
348 for several tens of thousands of years (2)) if they are to become established. However,
349 once established the survival of these flocs and the range of sizes observed suggest
350 that some flocs could migrate from these regions to further colonise the facility. There
351 is recent evidence (58) that microbial communities can reduce the ambient pH under
352 ILW conditions suggesting that the creation of low pH regions within an cementitious
353 GDF by microbial activity is possible. This would then provide initiation points from
354 which floc forming communities such as those described in this paper could propagate.

355 Overall our study demonstrates that in order to survive hyper-alkaline conditions
356 microorganisms are able to form multi species flocs composed of a complex mixture
357 of EPS which provides protection from alkaline pH values up to pH 13.0. The formation

358 of these flocs provides the microbial communities concerned with a means of
359 dispersion and propagation within hyper-alkaline environments such as a cementitious
360 GDF for ILW.

361 **Funding**

362 The work here was partially supported by both Radioactive Waste Management Ltd
363 (RWM Ltd) and the C14-BIG project (EPSRC, EP/I036354/1).

364 **Acknowledgements**

365 The Zeiss LSM880 microscope was purchased with a Wellcome Trust multi-user
366 equipment award: WT104918MA.

367 **References**

- 368 1. **NDA** 2013 Higher Activity Waste, The Long-term Management of Reactor Core
369 Graphite Waste Credible Options (Gate A), SMS/TS/D1-HAW-6/002/A. Nuclear
370 Decommissioning Authority (Radioactive Waste Management Directorate),
371 Harwell, Didcot, Oxfordshire, UK.
- 372 2. **NDA** 2010 Near-field Evolution Status Report., NDA/RWMD/033. Nuclear
373 Decommissioning Authority (Radioactive Waste Management Directorate),
374 Harwell, Didcot, Oxfordshire, UK.
- 375 3. **Chapman, N., and A. Hooper.** 2012. The disposal of radioactive wastes
376 underground. Proceedings of the Geologists' Association. **123**:46-63.

- 377 4. **Libert, M., O. Bildstein, L. Esnault, M. Jullien, and R. Sellier.** 2011.
378 Molecular hydrogen: An abundant energy source for bacterial activity in nuclear
379 waste repositories. *Physics and Chemistry of the Earth* **36**:1616-1623.
- 380 5. **Knill, C. J., and J. F. Kennedy.** 2003. Degradation of cellulose under alkaline
381 conditions. *Carbohydrate Polymers* **51**:281-300.
- 382 6. **Bassil, N. M., N. Bryan, and J. R. Lloyd.** 2014. Microbial degradation of
383 isosaccharinic acid at high pH. *The ISME journal* **9**:310-320.
- 384 7. **Rout, S. P., C. J. Charles, C. Doulgeris, A. J. McCarthy, D. J. Rooks, J. P.**
385 **Loughnane, A. P. Laws, and P. N. Humphreys.** 2015. Anoxic Biodegradation
386 of Isosaccharinic Acids at Alkaline pH by Natural Microbial Communities. *PLOS*
387 *ONE* **10**:e0137682.
- 388 8. **Rout, S. P., C. J. Charles, E. J. Garratt, A. P. Laws, J. Gunn, and P. N.**
389 **Humphreys.** 2015. Evidence of the generation of isosaccharinic acids and their
390 subsequent degradation by local microbial consortia within hyper-alkaline
391 contaminated soils, with relevance to intermediate level radioactive waste
392 disposal. *PLOS ONE* **10**.
- 393 9. **Charles, C., S. Rout, E. Garratt, K. Patel, A. Laws, and P. Humphreys.** 2015.
394 The enrichment of an alkaliphilic biofilm consortia capable of the anaerobic
395 degradation of isosaccharinic acid from cellulosic materials incubated within an
396 anthropogenic, hyperalkaline environment. *FEMS microbiology ecology*:fiv085.
- 397 10. **Dang, H., and C. R. Lovell.** 2016. Microbial surface colonization and biofilm
398 development in marine environments. *Microbiology and Molecular Biology*
399 *Reviews* **80**:91-138.

- 400 11. **Katharios-Lanwermeijer, S., C. Xi, N. Jakubovics, and A. Rickard.** 2014.
401 Mini-review: microbial coaggregation: ubiquity and implications for biofilm
402 development. *Biofouling* **30**:1235-1251.
- 403 12. **Flemming, H. C., and J. Wingender.** 2010. The biofilm matrix. *Nature Reviews*
404 *Microbiology* **8**:623-33.
- 405 13. **Droppo, I. G., G. G. Leppard, D. T. Flannigan, and S. N. Liss.** 1997. The
406 freshwater floc: A functional relationship of water and organic and inorganic floc
407 constituents affecting suspended sediment properties. *Water, Air, and Soil*
408 *Pollution* **99**:43-54.
- 409 14. **Davey, M. E., and G. A. O'toole.** 2000. Microbial biofilms: from ecology to
410 molecular genetics. *Microbiology and Molecular Biology Reviews* **64**:847-867.
- 411 15. **Melo, L.** 2005. Biofilm physical structure, internal diffusivity and tortuosity.
412 *Water Science & Technology* **52**:77-84.
- 413 16. **Jiao, Y., P. D'haeseleer, B. D. Dill, M. Shah, N. C. VerBerkmoes, R. L.**
414 **Hettich, J. F. Banfield, and M. P. Thelen.** 2011. Identification of biofilm matrix-
415 associated proteins from an acid mine drainage microbial community. *Applied*
416 *and Environmental Microbiology* **77**:5230-5237.
- 417 17. **Hallbeck, L.** 2009 Microbial processes in glaciers and permafrost. A literature
418 study on microbiology affecting groundwater at ice sheet melting, SKB Rapport
419 R-09-37. Report prepared for Svensk Kärnbränslehantering AB, Swedish
420 Nuclear Fuel and Waste Management Co, Stockholm, Sweden
- 421 18. **de Boer, J. P., C. C. Cronenberg, D. de Beer, J. C. van den Heuvel, M. J. T.**
422 **de Mattos, and O. M. Neijssel.** 1993. pH and glucose profiles in aggregates

- 423 of *Bacillus laevolacticus*. Applied and Environmental Microbiology **59**:2474-
424 2478.
- 425 19. **Lens, P. N., D. De Beer, C. C. Cronenberg, F. P. Houwen, S. P. Ottengraf,**
426 **and W. H. Verstraete.** 1993. Heterogeneous distribution of microbial activity in
427 methanogenic aggregates: pH and glucose microprofiles. Applied and
428 Environmental Microbiology **59**:3803-3815.
- 429 20. **Hunter, R. C., and T. J. Beveridge.** 2005. Application of a pH-sensitive
430 fluoroprobe (C-SNARF-4) for pH microenvironment analysis in *Pseudomonas*
431 *aeruginosa* biofilms. Applied and Environmental Microbiology **71**:2501-10.
- 432 21. **Babauta, J. T., H. D. Nguyen, T. D. Harrington, R. Renslow, and H. Beyenal.**
433 2012. pH, redox potential and local biofilm potential microenvironments within
434 *Geobacter sulfurreducens* biofilms and their roles in electron transfer.
435 Biotechnology and Bioengineering **109**:2651-62.
- 436 22. **Vangsgaard, A. K., M. Mauricio-Iglesias, B. Valverde-Perez, K. V. Gernaey,**
437 **and G. Sin.** 2013. pH variation and influence in an autotrophic nitrogen
438 removing biofilm system using an efficient numerical solution strategy. Water
439 Science and Technology **67**:2608-15.
- 440 23. **Allan, V., L. Macaskie, and M. Callow.** 1999. Development of a pH gradient
441 within a biofilm is dependent upon the limiting nutrient. Biotechnology Letters
442 **21**:407-413.
- 443 24. **Hurt, R., M. Robeson, M. Shakya, J. Moberly, T. Vishnivetskaya, B. Gu, and**
444 **D. Elias.** 2014. Improved yield of high molecular weight DNA coincides with

- 445 increased microbial diversity access from iron oxide cemented sub-surface clay
446 environments. PLOS ONE **9**:e102826.
- 447 25. **Griffiths, R. I., A. S. Whiteley, A. G. O'Donnell, and M. J. Bailey.** 2000. Rapid
448 Method for Coextraction of DNA and RNA from Natural Environments for
449 Analysis of Ribosomal DNA- and rRNA-Based Microbial Community
450 Composition. Applied and Environmental Microbiology **66**:5488-5491.
- 451 26. **Klindworth, A., E. Pruesse, T. Schweer, J. Peplies, C. Quast, M. Horn, and**
452 **F. O. Glockner.** 2013. Evaluation of general 16S ribosomal RNA gene PCR
453 primers for classical and next-generation sequencing-based diversity studies.
454 Nucleic Acids Research **41**:e1.
- 455 27. **Jung, M.-Y., S.-J. Park, D. Min, J.-S. Kim, W. I. C. Rijpstra, J. S. S. Damsté,**
456 **G.-J. Kim, E. L. Madsen, and S.-K. Rhee.** 2011. Enrichment and
457 characterization of an autotrophic ammonia-oxidizing archaeon of mesophilic
458 crenarchaeal group I. 1a from an agricultural soil. Applied and Environmental
459 Microbiology **77**:8635-8647.
- 460 28. **Schloss, P. D., S. L. Westcott, T. Ryabin, J. R. Hall, M. Hartmann, E. B.**
461 **Hollister, R. A. Lesniewski, B. B. Oakley, D. H. Parks, and C. J. Robinson.**
462 2009. Introducing mothur: open-source, platform-independent, community-
463 supported software for describing and comparing microbial communities.
464 Applied and Environmental Microbiology **75**:7537-7541.
- 465 29. **Kim, O. S., Y. J. Cho, K. Lee, S. H. Yoon, M. Kim, H. Na, S. C. Park, Y. S.**
466 **Jeon, J. H. Lee, H. Yi, S. Won, and J. Chun.** 2012. Introducing EzTaxon-e: a
467 prokaryotic 16S rRNA gene sequence database with phylotypes that represent

- 468 uncultured species. *International Journal of Systematic and Evolutionary*
469 *Microbiology* **62**:716-21.
- 470 30. **Chen, M.-Y., D.-J. Lee, J.-H. Tay, and K.-Y. Show.** 2007. Staining of
471 extracellular polymeric substances and cells in bioaggregates. *Applied*
472 *Microbiology and Biotechnology* **75**:467-474.
- 473 31. **Ainsworth, T., M. Fine, L. Blackall, and O. Hoegh-Guldberg.** 2006.
474 Fluorescence in situ hybridization and spectral imaging of coral-associated
475 bacterial communities. *Applied and Environmental Microbiology* **72**:3016-3020.
- 476 32. **Amann, R. I., B. J. Binder, R. J. Olson, S. W. Chisholm, R. Devereux, and**
477 **D. A. Stahl.** 1990. Combination of 16S rRNA-targeted oligonucleotide probes
478 with flow cytometry for analyzing mixed microbial populations. *Applied and*
479 *Environmental Microbiology* **56**:1919-1925.
- 480 33. **Manz, W., R. Amann, W. Ludwig, M. Vancanneyt, and K.-H. Schleifer.** 1996.
481 Application of a suite of 16S rRNA-specific oligonucleotide probes designed to
482 investigate bacteria of the phylum cytophaga-flavobacter-bacteroides in the
483 natural environment. *Microbiology* **142**:1097-1106.
- 484 34. **Manz, W., R. Amann, W. Ludwig, M. Wagner, and K.-H. Schleifer.** 1992.
485 Phylogenetic oligodeoxynucleotide probes for the major subclasses of
486 proteobacteria: problems and solutions. *Systematic and Applied Microbiology*
487 **15**:593-600.
- 488 35. **Roller, C., M. Wagner, R. Amann, W. Ludwig, and K.-H. Schleifer.** 1994. In
489 situ probing of Gram-positive bacteria with high DNA G+C content using 23S
490 rRNA-targeted oligonucleotides. *Microbiology* **140**:2849-2858.

- 491 36. **Monique, R., G. N. Elisabeth, P. Etienne, and L. Dominique.** 2008. A high
492 yield multi-method extraction protocol for protein quantification in activated
493 sludge. *Bioresource Technology* **99**:7464-71.
- 494 37. **Dubois, M., K. Gilles, J. Hamilton, P. Rebers, and F. Smith.** 1951. A
495 colorimetric method for the determination of sugars. *Nature* **168**.
- 496 38. **Bradford, M. M.** 1976. A rapid and sensitive method for the quantitation of
497 microgram quantities of protein utilizing the principle of protein-dye binding.
498 *Analytical Biochemistry* **72**:248-254.
- 499 39. **Bligh, E. G., and W. J. Dyer.** 1959. A rapid method of total lipid extraction and
500 purification. *Canadian Journal of Biochemistry and Physiology* **37**:911-917.
- 501 40. **Marshall, V. M., A. P. Laws, Y. Gu, F. Levander, P. Rådström, L. De Vuyst,**
502 **B. Degeest, F. Vaningelgem, H. Dunn, and M. Elvin.** 2001.
503 Exopolysaccharide-producing strains of thermophilic lactic acid bacteria cluster
504 into groups according to their EPS structure. *Letters in Applied Microbiology*
505 **32**:433-437.
- 506 41. **Leivers, S., C. Hidalgo-Cantabrana, G. Robinson, A. Margolles, P. Ruas-**
507 **Madiedo, and A. P. Laws.** 2011. Structure of the high molecular weight
508 exopolysaccharide produced by *Bifidobacterium animalis* subsp. lactis IPLA-R1
509 and sequence analysis of its putative eps cluster. *Carbohydrate Research*
510 **346**:2710-2717.
- 511 42. **B.S.I** 1999 BS 5766-8:1999 ISO 6496:1999, Methods for analysis of animal
512 feeding stuffs —Part 8: Determination of moisture and other volatile matter
513 content., British Standards Institute, London, UK

- 514 43. **B.S.I.** 2005. BS ISO 14853:2005 Plastics-Determination of the ultimate
515 anaerobic biodegradation of plastic materials in an aqueous system- Method
516 by measurement of biogas production. British Standards Institute, London, UK.
- 517 44. **Rout, S. P., J. Radford, A. P. Laws, F. Sweeney, A. Elmekawy, L. J. Gillie,**
518 **and P. N. Humphreys.** 2014. Biodegradation of the Alkaline Cellulose
519 Degradation Products Generated during Radioactive Waste Disposal. PLOS
520 ONE **9**:e107433.
- 521 45. **Jung, J., J. Chun, and W. Park.** 2012. Genome sequence of extracellular-
522 protease-producing *Alishewanella jeotgali* isolated from traditional Korean
523 fermented seafood. Journal of Bacteriology **194**:2097.
- 524 46. **Yumoto, I., A. Nakamura, H. Iwata, K. Kojima, K. Kusumoto, Y. Nodasaka,**
525 **and H. Matsuyama.** 2002. *Dietzia psychralcaliphila* sp. nov., a novel,
526 facultatively psychrophilic alkaliphile that grows on hydrocarbons. International
527 Journal of Systematic and Evolutionary Microbiology **52**:85-90.
- 528 47. **Koerner, R. J., M. Goodfellow, and A. L. Jones.** 2009. The genus *Dietzia*: a
529 new home for some known and emerging opportunist pathogens. FEMS
530 Immunology and Medical Microbiology **55**:296-305.
- 531 48. **Briley, K. A., L. B. Camilleri, G. M. Zane, J. D. Wall, and M. W. Fields.** 2014.
532 Biofilm growth mode promotes maximum carrying capacity and community
533 stability during product inhibition syntrophy. Frontiers in Microbiology **5**:693.
- 534 49. **Gorlenko, V., A. Tsapin, Z. Namsaraev, T. Teal, T. Tourova, D. Engler, R.**
535 **Mielke, and K. Nealson.** 2004. *Anaerobranca californiensis* sp. nov., an
536 anaerobic, alkalithermophilic, fermentative bacterium isolated from a hot spring

- 537 on Mono Lake. International Journal of Systematic and Evolutionary
538 Microbiology **54**:739-743.
- 539 50. **Sorokin, I. D., E. V. Zadorina, I. K. Kravchenko, E. S. Boulygina, T. P.**
540 **Tourova, and D. Y. Sorokin.** 2008. *Natronobacillus azotifigens* gen. nov., sp.
541 nov., an anaerobic diazotrophic haloalkaliphile from soda-rich habitats.
542 Extremophiles **12**:819-27.
- 543 51. **Janto, B., A. Ahmed, M. Ito, J. Liu, D. B. Hicks, S. Pagni, O. J. Fackelmayer,**
544 **T.-A. Smith, J. Earl, L. D. H. Elbourne, K. Hassan, I. T. Paulsen, A.-B.**
545 **Kolstø, N. J. Tourasse, G. D. Ehrlich, R. Boissy, D. M. Ivey, G. Li, Y. Xue,**
546 **Y. Ma, F. Z. Hu, and T. A. Krulwich.** 2011. Genome of alkaliphilic *Bacillus*
547 *pseudofirmus* OF4 reveals adaptations that support the ability to grow in an
548 external pH range from 7.5 to 11.4. Environmental Microbiology **13**:3289-3309.
- 549 52. **Perry IV, T. D., V. Klepac-Ceraj, X. V. Zhang, C. J. McNamara, M. F. Polz,**
550 **S. T. Martin, N. Berke, and R. Mitchell.** 2005. Binding of harvested bacterial
551 exopolymers to the surface of calcite. Environmental Science and Technology
552 **39**:8770-8775.
- 553 53. **Das, T., S. Sehar, L. Koop, Y. K. Wong, S. Ahmed, K. S. Siddiqui, and M.**
554 **Manefield.** 2014. Influence of calcium in extracellular DNA mediated bacterial
555 aggregation and biofilm formation. PLOS ONE **9**:e91935.
- 556 54. **Endo, T., K. Nakamura, and H. Takahashi.** 1976. Pronase-susceptible floc
557 forming bacteria: relationship between flocculation and calcium ion. Agricultural
558 and Biological Chemistry **40**:2289-2295.

- 559 55. **Enomoto, K., and N. Koyama.** 1999. Effect of growth pH on the phospholipid
560 contents of the membranes from alkaliphilic bacteria. *Current Microbiology*
561 **39**:270-3.
- 562 56. **Higgins, M. J., and J. T. Novak.** 1997. Characterization of exocellular protein
563 and its role in bioflocculation. *Journal of Environmental Engineering* **123**:479-
564 485.
- 565 57. **Tsuji, K.** 2002. Donnan equilibria in microbial cell walls: a pH-homeostatic
566 mechanism in alkaliphiles. *Colloids and Surfaces B: Biointerfaces* **24**:247-251.
- 567 58. **Bassil, N. M., A. D. Bewsher, O. R. Thompson, and J. R. Lloyd.** 2015.
568 Microbial degradation of cellulosic material under intermediate-level waste
569 simulated conditions. *Mineralogical Magazine* **79**:1433-1441.
- 570
- 571

572 **Table and Figure Legends**

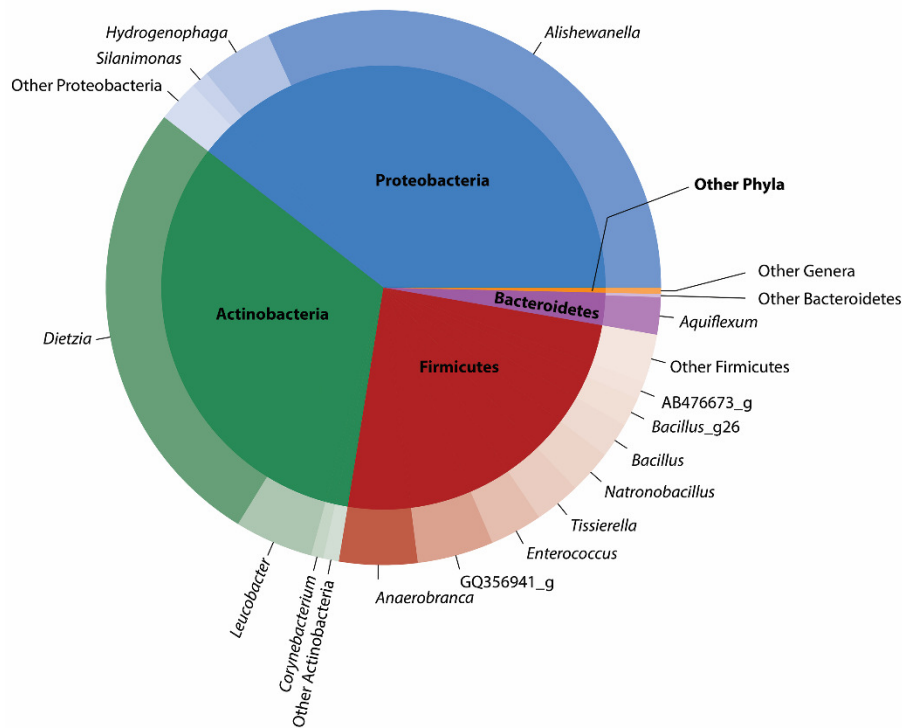
Sugar	Composition (%)
Mannose	50.53
Ribose	9.89
Ribitol	9.73
Arabinose	5.71
Rhamnose	5.07
Galactose	4.45
Trehalose	4.45
Glucuronic	3.51
Xylose	2.91
Fucose	2.00
Glucose	0.99
Galacturonic	0.76

573

574 **Table 1: Monomer composition of carbohydrates associated with floc EPS.**

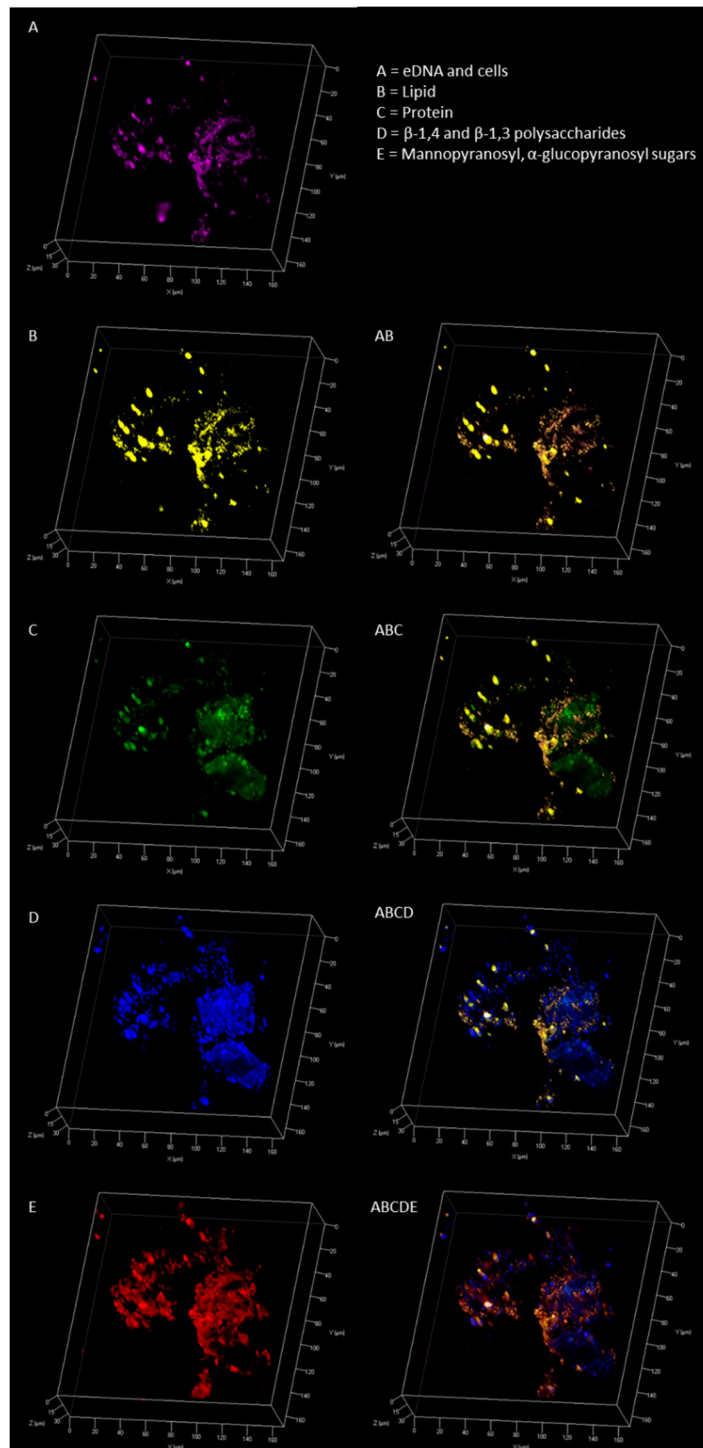
575 Mannose accounted for 50 % of the monomers present with ribose and ribitol making
 576 up a further 20 %. A varied range of sugars were identified in smaller amounts with
 577 uronic acids also present but only making up 4.3 % of the total monomer composition.

578



579

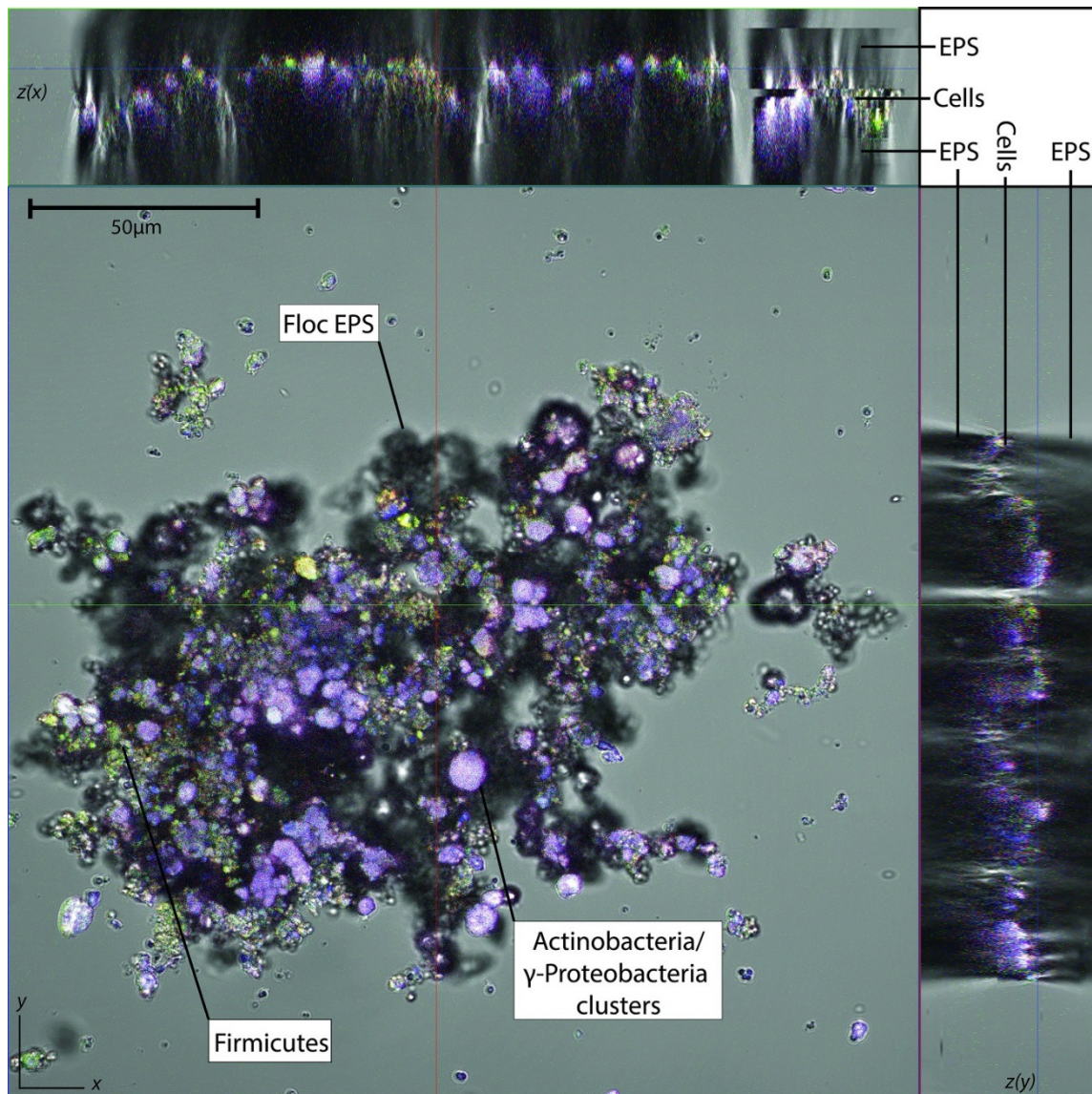
580 **Fig. 1: 16S rRNA gene profile of the microcosm community.** The microcosm
 581 community was dominated by sequence reads associated with the Phyla
 582 Proteobacteria (39.49 %), Actinobacteria (32.93 %), Firmicutes (24.85 %) and
 583 Bacteroidetes (2.34 %) making up 99.6 % of the library (Inner pie chart). The
 584 Proteobacteria and Actinobacteria were each dominated by sequence reads
 585 associated with *Alishewanella* (31.81 %) and *Dietzia* (26.74 %) respectively (Outer
 586 chart ring).



587

588 **Fig. 2: CLSM imaging of EPS components.** CLSM revealed the flocs to be
 589 composed of a complex mixture of proteins, carbohydrates, lipids, eDNA and cells.
 590 The most basal layer of the floc was composed primarily of lipids and β -1,4 and β -1,3
 591 polysaccharides, where the most outer layers of the floc were composed of
 592 concentrated areas of proteins, these regions were surrounded by α -mannopyranosyl
 593 and α -glucopyranosyl sugars which were also closely associated with eDNA. Images

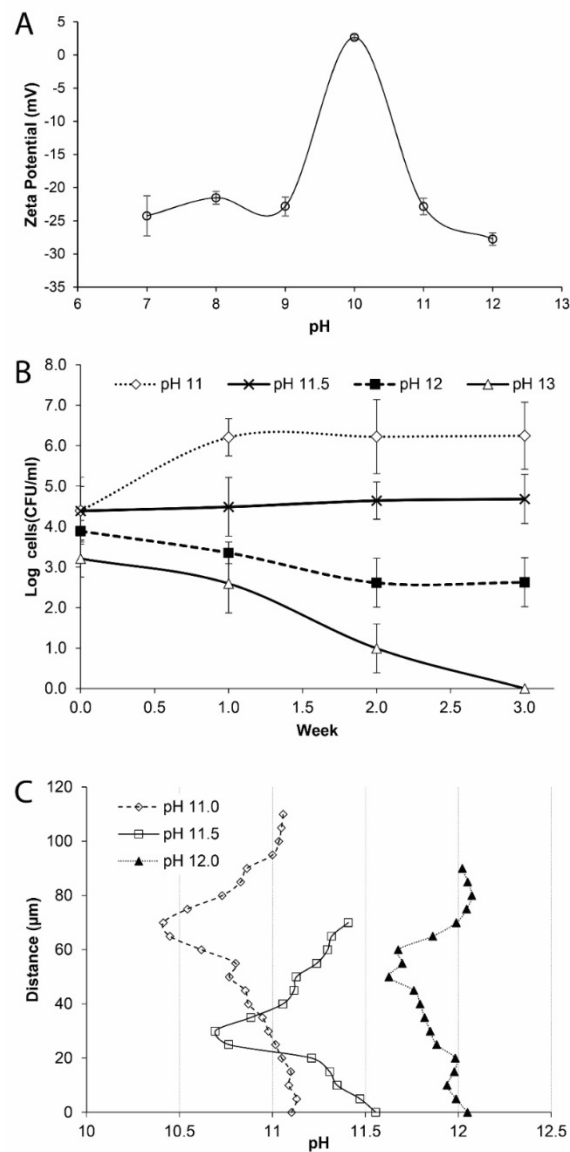
594 in the left column show the individual components of the EPS, images in the right hand
595 column show the composite images.



596

597 **Fig 3. FISH/CLSM imaging of the bacterial floc community.** The optical Z stack
598 shows the bacterial cells to be concentrated in the centre of the floc, surrounded by
599 ~10 µm of EPS material (z(y), z (x) images). FISH/CLSM probing showed that the
600 Actinobacteria (red) and γ-Proteobacteria (blue) were clustered together, with
601 Firmicutes (green) being situated to the periphery of these clusters. The flocs ranged
602 from 50 – 250 µm in diameter and most featured a central well defined mass with

603 looser less formed sections attached. The denser areas of the flocs showed higher
604 numbers of cells and more concentrated areas of all EPS components.



605

606 **Figure 4: Floc response to pH.** Analysis of the zeta potential of the flocs under a
607 range of pH values showed consistent stable potentials of -20 to -30 mV between pH
608 7-9 and 10-12 (Fig. 4A). Following exposure to elevated pH values flocs demonstrated
609 the ability to grow at pH 11.0, survive for >3 weeks at pH 12.0 and 2 weeks at pH 13.0
610 (Fig. 4B). Profiling of the pH indicated that the interior of the flocs had a lower pH value
611 than the exterior pH when exposed to pH 11.0, 11.5 and 12.0 indicating pH shifts of

612 0.6, 0.8 and 0.4 pH units. These pH shifts occurred over short distances with the
613 largest pH shifts occurring over a distance of 35 μ m (Fig. 4C).

614

615

616

617