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1 **Biochemical composition and changes of extracellular polysaccharides**  
2 **(ECPS) produced during microphytobenthic biofilm development**  
3 **(Marennes-Oléron, France).**

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11

12 **Abstract**

13 The main goal of this work was to study the dynamics and biochemical composition of  
14 ExtraCellular Polysaccharides (ECPS), a fraction of the Extracellular Polymeric Substances  
15 (EPS) produced during the development of a microphytobenthic biofilm in a European  
16 intertidal mudflat (Marennes-Oléron Bay, France) during winter. Microphytobenthic biomass  
17 was surveyed during four consecutive emersion periods to confirm the biofilm growth.  
18 Bacteria abundance was also checked considering the importance of heterotrophic bacteria  
19 observed by various authors in the dynamics of EPS. Various colorimetric assays, coupled to  
20 biochemical chromatographic analysis were used to characterize the three main fractions of  
21 extracted EPS: colloidal, bound and residual. The monosaccharide distribution of colloidal  
22 ECPS highlighted their role of carbon source for bacteria (> 50 % of glucose) even if no  
23 increase of colloidal carbohydrate amounts was observed during the tidal exposure. Bound

24 ECPS were composed of deoxy or specific sugars (30 % rhamnose) and uronic acids (18 %  
25 galacturonic acid). Their levels and dynamics could be correlated to the development of the  
26 microphytobenthic biofilm, enhancing the stabilization of the sediment or increasing binding  
27 forces accordingly. Residual fractions, containing refractory bound ECPS and other internal  
28 polymeric substances, were composed of various carbohydrates. The high ratio of glucose in  
29 these fractions (18 to 43 %) was interesting as it was once attributed to colloidal sugars due to  
30 poor extraction procedures. Finally, the presence of inositol (15 %) was significant since no  
31 author has highlighted it before, knowing that inositol is a major growth factor for  
32 heterotrophic bacteria.

33 *Key words:* extracellular compounds, EPS, biochemical characterization, biofilm, benthic  
34 ecology, intertidal mudflat

35

## 36 **Introduction**

37 Tidal flats are an essential system within the trophic network and contribute greatly to the  
38 productivity of coastal ecosystems. The high primary and secondary productivity of mudflats  
39 is linked to physical and biological processes involving unicellular micro-organisms. The  
40 development of conceptual models for these highlighted the role of diatoms [16] and bacteria  
41 in the sediment mudflats [21]. More generally, regarding biomass and production, it is known  
42 that microphytobenthos are a major component of these intertidal sediment communities [38,  
43 39, 41].

44 Diatoms have been described in several recent studies showing that they were closely  
45 involved in the formation of biofilms [11, 27]. They are essential for the health of the marine  
46 environment through the excretion of large quantities of Extracellular Polymeric Substances  
47 (EPS), which are involved in particular in their motility systems. In intertidal benthic

48 ecosystems, this motility is essential to enable epipelagic diatoms to migrate into the photic zone  
49 of the sediment during the time of emersion [39]. In addition, previous research has shown  
50 that EPS can be used as a carbon source by the bacterial community and the macrofauna [17].  
51 EPS also affect the microenvironment of biofilms by varying physico-chemical parameters  
52 like porosity, density, sorption properties, hydrophobicity and mechanical stability [13, 14].  
53 Thus, the composition of EPS is clearly linked to the erosion of sediments, either increasing  
54 their stability [26] or decreasing stability in case of nutrient stress during growth stationary  
55 phase [25].

56 The microbenthic community is mostly composed of water, diatoms, bacteria, eukaryotic  
57 microbes, EPS and inorganic particles. According to Wingender et al. (1999), the EPS matrix  
58 is composed of a wide variety of proteins, polysaccharides (ECPS), lipids and other more  
59 surprising compounds such as nucleic acids. This very complex composition makes  
60 biochemical analysis particularly difficult due to the diversity in sugar monomers, linkages,  
61 low concentrations and the interactions of compounds during assays. Besides, the extraction  
62 of EPS is another challenge since the polymers are highly complex and exist in a continuum  
63 from tightly cell bound to loosely cell associated. The extraction and separation methods have  
64 to be selected in term of the compounds to be extracted while controlling what happens to  
65 other compounds; minimal cell lysis and no disruption or alteration of the EPS are basic  
66 requirements [9]. In this way, Takahashi et al. (2009) clearly demonstrated that extraction of  
67 colloidal and bound EPS with a cation exchange resin was the best method to obtain high  
68 yield of carbohydrates and a proper extraction without causing cell disruption or  
69 contamination by internal storage materials.

70 Extraction protocols can distinguish a range of EPS types, depending on the nature of the  
71 extraction and the location of EPS in the sediment [6] such as: (i) colloidal fractions  
72 corresponding to the polymeric substances excreted in the medium, (ii) bound fractions which

73 are the compounds surrounding the cells (that should be associated to cell adhesion and/or  
74 protection) and (iii) residual fractions, corresponding to the internal polymers and refractory  
75 bounds EPS [37]. It has been demonstrated that diatom and microbial EPS can be also  
76 separated into two major types: Low Molecular Weight (LMW) and High Molecular Weight  
77 (HMW).

78 The present investigation used *in situ* an improved extraction method involving a cation  
79 exchange resin to better characterize the EPS and particularly ECPS secreted by a  
80 microphytobenthic biofilm [37]. The three fractions of EPS were extracted from intertidal  
81 sediments (Marennes-Oléron Bay, France), during the development of diatom-dominated  
82 biofilm [18, 19, 25] and three consecutive diurnal emersion periods during winter. The main  
83 goal of the study was to determine and understand, through biochemical analysis of these  
84 fractions, the dynamics of ECPS during a microphytobenthic biofilm growth.

## 85 **Materials and methods**

### 86 *Intertidal mudflat samples*

87 The mudflat samples used in this study were collected from Marennes-Oléron Bay (Atlantic  
88 Coast of France), during one week in February 2008 (winter) at low tide (Fig. 1). The field  
89 sampling was organized as a chessboard where we defined square sampling units (2m-side)  
90 separated by alleys (2m in width). Every day, 3 squares were randomly taken to account for  
91 spatial heterogeneity. Three cores of each selected square were sampled every hour during the  
92 emersion period, for 3 consecutive days. Sediment samples from each square were collected  
93 by using core diameter of 20 cm. For each core, the top 1cm was collected three times and  
94 pooled. After each sampling, sediment was brought back from the field by using a watercraft  
95 for an immediate EPS extraction on fresh sediments on the upper shore. Biochemical analyses  
96 were performed in triplicate on the colloidal, bound and residual fractions (216 fractions).

97 *Environmental parameters*

98 Light was measured using a Li-Cor sensor which was recorded every minute during sampling  
99 days. Enumeration of bacteria: to separate bacteria from sediment particles, incubation in  
100 pyrophosphate (0.01 M for at least 30 min) and sonication (60 W) were performed. Bacteria  
101 from sediment were labeled using 4,6-diamidino-2-phenylindole dihydrochloride (DAPI)  
102 ( $2500 \mu\text{g} \cdot \text{l}^{-1}$ ), filtered onto 0.2  $\mu\text{m}$  Nucleopore black filter [30] and then counted by  
103 microscopy (x 1000, Axioskop, Zeiss). The chlorophyll *a* concentration in the sediment was  
104 measured using fluorometry method [23] on two types of samples which were obtained from  
105 the top 0.2 cm (more representative of the chlorophyll *a* produced by diatoms) or 1 cm of  
106 sediment cores. Initially, 16 measures of chl *a* concentration were planned but only 9  
107 measures were done in the top 1 cm of sediment cores (from the day 2/18/08 to the day  
108 2/20/08) and 12 measures were done in the top 0.2 cm (from the day 2/18/08 to the day  
109 2/21/08). Missing data were due to experiment constraints.

110 *Materials*

111 Dowex Marathon C, bichinchonic acid (BCA) Protein Assay Kit, Azure A, N,O-  
112 bis(trimethylsilyl)trifluoroacetamide: trimethylchlorosilane (BSTFA: TMCS) (99:1) were  
113 obtained from Sigma-Aldrich. Standard carbohydrates (dextran, dextran sulfate, heparin,  
114 fucoidan, glucose, galactose, rhamnose, fucose, fructose, xylose, arabinose, mannose, *myo*-  
115 inositol, glucuronic and galacturonic acid) and a protein standard (Bovine Serum Albumin,  
116 BSA) were obtained from Sigma-Aldrich. The DB-1701 JandW Scientific column (30 m, 0.32  
117 mm, 1  $\mu\text{m}$ ) for Gas Chromatography-Mass Spectrometry analysis (GC/MS) was obtained  
118 from Agilent.

119 *EPS Extraction*

120 The extraction method [37] was done immediately after sampling and sediment mixing in the  
121 field. 20 mL of fresh mudflat was continuously mixed with 20 mL of Artificial Sea Water  
122 (ASW 30 Practical Salinity Units) during 1 h in darkness at 4 °C and then centrifuged at 3500  
123 g and 4 °C for 10 min. The supernatant (a) containing colloidal EPS was collected and stored  
124 at 4 °C. 20 mL of ASW and 1 g of activated Dowex (Marathon C, activated in Phosphate  
125 Buffer Saline for 1 h in the dark) was added to the sediment pellet (b). The samples were  
126 mixed gently at 4 °C for 1 h in the dark and then centrifuged at 3500 g and 4 °C for 10 min. A  
127 supernatant containing the bound EPS (c) and a cap containing intracellular and residual  
128 polymers (d) were obtained. The cap was then frozen. The residual polymers were extracted  
129 from the frozen sediment samples (d), by sonication at 100 W for 3 min on ice after  
130 resuspension in 20 mL in ASW.

131 For each fraction (colloidal, bound and residual polymers), absolute ethanol at -20 °C was  
132 added to the sample (a) to obtain a final ethanol concentration of 75 % (v/v). The solution was  
133 gently mixed and stored overnight at -20 °C. The solution was then centrifuged at 3500 g and  
134 4 °C for 15 min to obtain a supernatant (Low Molecular Weight, LMW fraction) and a  
135 precipitate pellet (High Molecular Weight, HMW fraction). Finally, the fractions were dried  
136 under air flow and stored at -20 °C.

#### 137 *Biochemical analysis of EPS and carbohydrate fractions*

138 Total sugar content was determined using the phenol-sulfuric acid assay, using glucose as a  
139 standard [10]. Briefly, 1 mL of 5 % phenol and 5 mL sulfuric acid were added to 1 mL EPS  
140 solution, previously resuspended in ultra-pure water, and vortexed. Measurements were read  
141 after 30 min with a spectrophotometer at 485 nm. Total sugar amounts for the fractions were  
142 measured and normalized to chlorophyll *a* (chl *a*). This normalization is classically used in the

143 literature and allows to overestimates diatom EPS and ECPS production, comparing to other  
144 EPS sources, especially when the mudflat is mainly composed of microphytobenthos [18].

145 Protein content was determined using the bicinchoninic acid (BCA) assay, using bovine  
146 serum albumin (BSA) as a standard [31]. Briefly, 0.2 mL EPS, previously resuspended in  
147 ultra-pure water, were added to 2 mL BCA active reagent, and gently vortexed.  
148 Measurements were read after 15 min of heating at 60 °C, with a spectrophotometer at 562  
149 nm.

150 Uronic acid content was determined using the meta-hydroxydiphenyl method (MHDP), using  
151 galacturonic and glucuronic acids as standards [3, 12]. Briefly, 40 µL of 4 M sulfamic acid  
152 and 2.4 mL of 0.075 M sodium tetraborate were added to 400 µL of EPS, previously  
153 resuspended in ultra-pure water, and gently vortexed. 80 µL of 0.15 % MHDP were added to  
154 the solution after 20 min of heating at 80 °C. Sample was strongly vortexed and allowed to  
155 rest 10 min. Measurements were read with a spectrophotometer at 525 nm.

156 The sulfate content was measured by the Azure A [22] and the Ba/Cl<sub>2</sub> gelation method [7],  
157 using Dextran sulfate as a standard. Concerning the Azure A assay, 2 mL of 10 mg/L Azure A  
158 were added to 200 µL EPS, previously resuspended in ultra-pure water, and gently vortexed.  
159 Measurements were read with a spectrophotometer at 535 nm. Concerning the Ba/Cl<sub>2</sub> gelation  
160 assay, 10 mg EPS were hydrolyzed in 0.5 mL of 2 M HCl for 2 h at 100 °C. Ultrapure water  
161 was added (qsp 10 mL) and the sample was then centrifuged for 10 min at 5000 g. For the  
162 turbidimetric assay, 1 mL supernatant, 9 mL ultra-pure water, 1 mL of 0.5 N HCl and 0.5 mL  
163 of Ba/Cl<sub>2</sub> – gelatin reagent were added in a glass tube. After 30 min resting at room  
164 temperature, measurements were read with a spectrophotometer at 500 nm.

165 *Characterization of carbohydrate fractions by GC/MS*

166 Prior to carbohydrate characterization by GC/MS, EPS fractions were solubilized in 5 mL of  
167 ultrapure water, dialyzed (6-8 KDa) and freeze-dried [2]. EPS were then dissolved in 2 M  
168 HCl at 50 mg/mL and heated at 90 °C for 4 h. The preparation (which contained mostly ECPS  
169 monomers) was then freeze-dried and stored at -20 °C. Analysis of the carbohydrate fractions  
170 were carried out by GC/MS using a Varian CP-3800 GC/Varian Saturn 2000. 400 µL of  
171 pyridine and 400 µL of BSTFA: TMCS (99: 1) was added to 2 mg of purified  
172 monosaccharides. The solution was mixed for 2 h at room temperature, then injected into a  
173 DB-1701 J&W Scientific column (30 m, 0.32 mm, 1 µm) at a flow of 1 mL/min. The helium  
174 pressure was 8.8 psi. The temperature of the injector was set at 250 °C. The rise in  
175 temperature in the oven was programmed for a first step at 150 °C for 0 min, then an  
176 increment of 10 °C/min up to 200 °C with a final step at 200 °C for 35 min. The ionization  
177 was performed by Electronic Impact (EI, 70 eV), the trap temperature was set at 150 °C and  
178 the target ion was fixed at 40-650 m/z [29]. Each monosaccharide amount measured by  
179 GC/MS was expressed depending on the total carbohydrate amounts measured by  
180 biochemical assays, while considering the sampling date and the emersion time.

### 181 *Statistical analysis*

182 One-way analysis of variance (ANOVA), was used to analyze changes in carbohydrate and  
183 uronic acid amounts among abiotic parameters (sampling location, emersion time) for each  
184 day. Data transformations (root) were performed each time it was required to check  
185 application conditions (normality). Normality tests of residuals were tested by Kolmogorov-  
186 Smirnov test and homoscedasticity by Bartlett test. In addition, post hoc procedures (Tukey  
187 test) were performed to analyze pairwise differences. Pearson correlations were performed to  
188 investigate the relationships between the different EPS fractions with biotic parameters  
189 (bacterial density, chl *a* biomass) and also abiotic parameters (luminosity). Statistical analyses  
190 were run using the statistical software XLStat (Addinsoft).

## 191 **Results**

### 192 Environmental parameters

193 Light measurements showed the higher brightness during the first day of sampling (1000  
194  $\mu\text{mol photons} \cdot \text{m}^{-2} \cdot \text{s}^{-1}$ ) (Fig. 2A). The most important brightness peaks were obtained for the  
195 other days around 12-14h (600 to 800  $\mu\text{mol photons} \cdot \text{m}^{-2} \cdot \text{s}^{-1}$ ). The comparison between  
196 ambient light climate and carbohydrate concentrations did not reveal a specific impact of light  
197 on the dynamics of ECPS except for bound HMW ( $r = -0.444$ ,  $R^2 = 0.197$ ). The measures of  
198 chlorophyll *a* remained constant during the sampling period, with values around 21.5  $\mu\text{g/g}$  of  
199 dry sediment (Fig. 2B). The survey showed the presence of a biofilm and its formation (Fig.  
200 2C). A significant increase of the Chl *a* concentration was observed for the day 2/21/08 (P  
201 value [5 %] = 0.003) (Fig. 2C).

### 202 Carbohydrate content

203 ECPS amounts and their dynamics for the three fractions are presented in Fig. 3, depending  
204 on the emersion time and the sampling day. The quantities of total colloidal carbohydrates  
205 (LMW and HMW) were close to 6.5  $\mu\text{g sugar} \cdot \mu\text{g chl } a^{-1}$  (Fig. 3A). The results showed that  
206 the amounts of LMW total sugars remained constant around 3.5  $\mu\text{g sugar} \cdot \mu\text{g chl } a^{-1}$  during  
207 the sampling campaign (except for 2/20/08-4h, with a concentration close to 5.5  $\mu\text{g sugar} \cdot \mu\text{g}$   
208  $\text{chl } a^{-1}$ ). HMW total sugar quantities were slightly lower (3  $\mu\text{g sugar} \cdot \mu\text{g chl } a^{-1}$  on average,  
209 with an increase for 2/20/08-4h to 5  $\mu\text{g sugar} \cdot \mu\text{g chl } a^{-1}$ ). Differences in concentrations  
210 related to the sampling squares were found but were not significant (p values [5 %] = 0.970  
211 and 0.121 for HMW and LMW colloidal carbohydrates respectively). No significant effect of  
212 the sampling day was observed on the dynamic of LMW and HMW colloidal ECPS (p value  
213 [5 %] = 0.447 and 0.471). Statistical analysis showed that the dynamics of LMW and HMW  
214 bound carbohydrates varied with the sampling day (p value [5 %] = 0.001 and 0.023

215 respectively) (Fig. 3B). Besides, no significant effect of the spatial heterogeneity was found  
216 for LMW and HMW bound carbohydrate concentrations (p values [5 %] = 0.957 and 0.324  
217 respectively). The LMW total sugar quantities in bound fractions tended to increase during  
218 the emerged phase. This trend was especially pronounced for the period 2/20/08 1 h - 4 h  
219 since the quantity nearly doubled (6  $\mu\text{g}$  to 13  $\mu\text{g}$  sugar .  $\mu\text{g chl } a^{-1}$ ) (p value for day 2/20/08 [5  
220 %] = 0.028). Concerning the residual fractions (Fig. 3C), there was a good level of  
221 repeatability for the LMW carbohydrate amounts between the sampling squares since no  
222 significant difference was found (p value [5 %] = 0.556). It is difficult to highlight a particular  
223 trend, except a stagnation of LMW carbohydrate amounts (around 20 $\mu\text{g}$  sugar .  $\mu\text{g chl } a^{-1}$ ).  
224 The amounts of HMW carbohydrate quantities in the residual fractions were found to increase  
225 but not significantly during the emersion of the sediment (p values for days 2/18-19-20/08 [5  
226 %] = 0.576, 0.976 and 0.057 respectively), at higher concentrations (30 to 75  $\mu\text{g}$  sugar .  $\mu\text{g}$   
227 chl  $a^{-1}$ ).

228 The change in distribution of total carbohydrates between LMW and HMW for the colloidal,  
229 bound and residual fractions is presented in Fig. 4. The LMW colloidal carbohydrates were  
230 slightly and significantly more abundant than the HMW colloidal carbohydrates (p value [5  
231 %] = 0.003). For the bound fractions, the carbohydrates were produced in very precise  
232 quantities as indicated by the low standard deviation between the different sampling sites. The  
233 bound carbohydrates seem to be produced at a constant and significant ratio of 90/10:  
234 LMW/HMW (p value [5 %] < 0.0001), compared with 55/45 for the colloidal fractions. The  
235 residual fractions were essentially composed of HMW compounds, *i.e.* 60-70 % for total  
236 carbohydrates.

237 Sulfate and protein contents

238 No sulfated groups were detected in the colloidal and bound fractions. Very low  
239 concentrations of proteins were found in the bound fractions. However, the BCA method used  
240 for analysis was not enough accurate, so these concentrations were considered close to zero.  
241 Sulfated components were detected in the residual fractions (Table 1). These amounts were  
242 relatively constant. However, these data should be interpreted with precaution in view of the  
243 significant deviations obtained between the different sampling sites. Protein concentrations  
244 were also measured in the residual fractions (Table 1). The quantities of proteins remained  
245 constant around  $20 \mu\text{g} \cdot \mu\text{g chl } a^{-1}$  throughout the emersion.

#### 246 Changes in ECPS composition

247 The changes in ECPS composition were carried out by GC/MS analysis (Fig. 5). The LMW  
248 colloidal fractions (Fig. 5A) contained about  $\pm 23$  % uronic acids (80 % galacturonic acid and  
249 20 % glucuronic acid). These levels confirmed the results obtained by colorimetric assays.  
250 The LMW colloidal fractions contained more than 50 % glucose but also 5 % rhamnose, 10 %  
251 xylose and 7 % inositol (as myo-inositol). The percentage of rhamnose increased during the  
252 emerged periods (0 to 6.75 %). In contrast, the percentage of mannose and xylose decreased  
253 slightly during the four hours of emersion (12 % to 8.25 % and 5 % to 1 % respectively). For  
254 the HMW colloidal fractions, the percentage of uronic acids was lower than in the LMW  
255 fractions and decreased during the emerged periods (12 % to 7.7 %). The HMW colloidal  
256 fractions did not contain rhamnose. Concerning the monosaccharide composition of the bound  
257 fractions, one example of a spectrum analysis is presented in Fig. 6. The LMW bound  
258 fractions (Fig. 5B) were rich in rhamnose (20.8 % to 32.9 % during the emerged periods),  
259 xylose ( $\pm 24$  %), glucose ( $\pm 21$  %) and galacturonic acid. Small amounts of arabinose and  
260 mannose were also detected in the LMW bound fractions. The level of rhamnose greatly  
261 increased during the emerged periods. In contrast, the percentage of galacturonic acid ratios  
262 decreased significantly between the first and last hour of emersion (16 % to 6.5 %). For the

263 HMW fractions, rhamnose represented about 35 % of the total carbohydrates. The percentage  
264 of xylose was similar in LMW and HMW bound fractions ( $\pm 25$  %). The percentage of  
265 galacturonic acid remained constant in HMW fractions at around 16 % during the emersion.  
266 The monosaccharide composition of residual fractions was different compared to colloidal  
267 and bound fractions (Fig. 5C). The concentration of each monosaccharide varied widely  
268 during the emerged periods, making it difficult to define a typical composition. For the LMW  
269 fractions, the amount of rhamnose was relatively constant during the first hours of emersion  
270 ( $\pm 10$  %) and decreased drastically to 1 % during the last hour. A similar observation can be  
271 made for the percentage of xylose in these fractions (10 % to 0.7 %). In addition, the  
272 percentage of glucose followed an amazing trend during the emerged periods, decreasing  
273 slightly during the first three hours of emersion (18 % to 12 %) and dramatically increasing  
274 during the last hour to a level close to 43 % or almost half of the total monosaccharides.  
275 Similar observations were made for the HMW fractions, particularly regarding the changing  
276 levels of rhamnose (11 to 5 %) and glucose (31 to 13 % during the first three hours then an  
277 increase to 59 % during the fourth hour). Finally, the increase in the levels of unknown peaks  
278 suggests that the LMW and HMW residual fractions are really complex.

#### 279 ECPS dynamics and bacteria abundance

280 The sediment was more rich in bacteria during the day 2/19/08 (around  $5e^{+9}$  cells  $\cdot g^{-1}$  dry  
281 sediment) (Fig. 2B). The day 2/18/08 showed the lowest concentrations of bacteria ( $1e^{+9}$  cells  
282  $\cdot g^{-1}$  dry sediment). A decrease in the amount of bacteria was observed during the 2/20/08  
283 ( $5e^{+9}$  to  $2.5e^{+9}$  cells  $\cdot g^{-1}$  dry sediment), with significant differences found for the bacterial  
284 densities at 0h, 1h and 3h of emersion (P value [5 %] = 0.013). During this period, a strong  
285 increase in bound LMW carbohydrates in the sediment was observed (Fig. 2B). Nevertheless,  
286 no significant correlation was found between bacteria and bound LMW carbohydrates ( $r = -$

287 0.21,  $R^2 = 0.044$ ), bound HMW carbohydrates ( $r = 0.186$ ,  $R^2 = 0.035$ ) and more generally  
288 bound (LMW + HMW) carbohydrates ( $r = -0.17$ ,  $R^2 = 0.029$ ).

## 289 **Discussion**

### 290 Methodological limitation

291 Overall, 1g of dry sediment was composed of about 1600 to 1900  $\mu\text{g}$  of total carbohydrates  
292 and about 300 to 600  $\mu\text{g}$  of total proteins, without normalization to Chl *a*. Although a large  
293 portion was unknown, authors have considered that natural mudflats samples may contain  
294 large quantities of impurities [8, 40] and it has been proved that the phenol sulfuric acid assay  
295 was not 100 % efficient for measuring complex polysaccharides, causing an underestimation  
296 of the total quantities of EPS [21]. Previous results obtained at the same station in 1998  
297 showed that 1 g of sediment contained 700 to 1100  $\mu\text{g}$  of carbohydrates [35]. These authors  
298 have worked on the first 5mm of sediment, which represent more accurately the  
299 microphytobenthos biofilm. Our “first 10 mm approach” could overestimate diatom EPS  
300 production due to the contamination from other sediment EPS sources [28].

### 301 ECPS dynamics during the biofilm development process

302 The ECPS dynamics in relation to the microphytobenthic biofilm development were  
303 investigated. A microphytobenthic biofilm development was confirmed (Fig. 2B, 2C) during  
304 the sampling period and study conditions (Fig. 2A). One main trend of constitution was  
305 observed during the biofilm development process.

306 The rate of EPS production by diatom biofilms during tidal exposure periods is affected by  
307 endogenous rhythms, light levels and nutrient status [17, 40]. Given the current literature,  
308 numerous authors explained the need to understand the biochemistry and physical nature of  
309 EPS to link their carbohydrate composition with their ability to become an integral part of the

310 biofilm matrix owing to their physico-chemical properties [1, 4]. Colloidal carbohydrates  
311 were consistently produced during the three days of analysis (Fig. 3A). No particular trend  
312 was found during the emerged periods, in contrast to the results obtained by Taylor et al.  
313 (1999) which showed an increase in colloidal carbohydrate levels towards the end of the  
314 emerged periods. It is noteworthy that Taylor worked on EPS fractions containing colloidal  
315 EPS and internal polymers. LMW bound carbohydrate amounts strongly increased towards  
316 the end of the emerged period (Fig. 3B) in contrast to HMW bound carbohydrates. Many  
317 authors showed an increase in bound carbohydrate amounts during the tidal exposure period  
318 [17, 32, 38]. Bound fractions were composed of 90 % LMW carbohydrates and this  
319 percentage was extremely constant throughout the sampling period (Fig. 5B). This high ratio  
320 of LMW carbohydrates could be attributed to the acidic properties of the Dowex which could  
321 hydrolyze a part of HMW ECPS, thus producing an overestimation of LMW carbohydrates.  
322 Cell-cell and cell-substratum interactions are facilitated by the production of tightly bound  
323 carbohydrates [4, 42] through the presence of uronic acids (Table 1). The anionic properties  
324 of bound ECPS, conferred by uronic acids, could allow association of divalent cations,  
325 providing greater binding forces during the biofilm development process [36].

326 Residual fractions, composed of refractory bound ECPS, internal polymers and all sort of  
327 sources of ECPS, were rich in carbohydrates, exceeding six or seven times the colloidal and  
328 bound carbohydrate quantities (Fig. 3C). In contrast with other works, the abundance in  
329 carbohydrates did not increase during the daytime emersion period. Indeed, photoassimilated  
330 carbons, attributed to glucan and/or chrysolaminaran, could be stored in diatoms to provide  
331 energy during periods of darkness [39]. Otherwise, the results showed the presence of proteins  
332 in residual fractions (Table 1) which could be use for additional C-stores and/or structuring  
333 microalgae cells (glycoproteins) [5, 37].

334 Changes in ECPS composition and ecological relevance

335 Considering the low standard deviations found between our true replicates (< 5 %), it can be  
336 considered that the type of monosaccharides, their turnover, and their amounts were  
337 controlled during short-term emerged periods by complex systems involving environmental  
338 conditions, sediment erosion or the transport and deposition cycle [40]. The monosaccharide  
339 composition was heterogeneous between the different EPS fractions (Fig. 5) [34]. Colloidal  
340 ECPS were mainly composed of glucose, galacturonic acid, xylose and inositol (Fig. 5A).  
341 Bound EPS had a more balanced distribution of monosaccharide components: rhamnose,  
342 xylose, glucose and galacturonic acid (Fig. 5B). Residual fractions were composed of glucose,  
343 mannose, rhamnose, galactose and galacturonic acid (Fig. 5C). The changes in EPS  
344 composition during the biofilm development process were compared to previous works which  
345 classically extracted multiple EPS: (colloidal EPS, colloidal extract (cEPS), EPS from hot  
346 water extraction (HW) and EPS from hot bicarbonate extraction (HB)). The results were then  
347 adapted to the conceptual model of Underwood and Paterson (2003) and a discussion was  
348 proposed, based on Fig. 7.

349 At this point, it is noteworthy to declare that it was surprising that fucose, a recurrent deoxy  
350 sugar found in the literature, has not been highlighted although the GC-MS method allowed  
351 its detection. It was possible that the lack of fucose was linked to the environmental  
352 conditions, the physiological state of the microphytobenthic biofilm and the period of the  
353 sampling campaign (winter).

354 The becoming of colloidal ECPS: are they only used as a C-source for bacteria?

355 LMW colloidal fractions could be compared to the low molecular weight exudates of  
356 Underwood and Paterson (2003), the colloidal fractions of Bellinger et al. (2009) or Hofmann  
357 et al. (2009). These exudates were mainly composed of glucose (53 %). HMW colloidal  
358 fractions (Fig. 7) could be compared to the category of colloidal EPS [40] or cEPS of

359 Abdullahi et al. (2006) and Hofmann et al. (2009). Carbohydrate content was mainly  
360 composed of glucose (55 %) (Fig. 5). Numerous studies have highlighted the role of bacteria  
361 in the dynamics of EPS on sediments [17, 21]. To better understand colloidal EPS and ECPS  
362 dynamics during the biofilm development process, it is also important to focus on these  
363 bacteria. Literature showed that microorganisms were able to produce specific bacterial EPS  
364 and to consume polymeric substances, as ECPS, for their own development. In this way,  
365 recent studies showed a decrease of LMW and HMW colloidal carbohydrates due to bacterial  
366 consumptions [21]. The present investigation did not show such a decrease (Fig. 2B) even if  
367 our specific sugar distribution of LMW colloidal fractions showed amounts of monomers  
368 close to those obtained by many authors, with the prevalence of glucose (Fig. 5). It is  
369 noteworthy that other monosaccharides could be involved in this consumption phenomenon.  
370 The percentages of hexoses (xylose, mannose) decreased during the emerged periods,  
371 suggesting that they were may be more easily assimilated or used by heterotrophic bacteria.  
372 Nonetheless, the important levels of xylose and mannose could be due to physiological  
373 processes to enhance the assimilation of specific proteins or molecules in cells [20]. The  
374 composition of colloidal ECPS could suggest that they were not only used as a nutrient  
375 source. In this way, the high level of galacturonic acid (> 12 %) and its decrease during the  
376 biofilm development was intriguing. Colloidal uronic acids could be involved in the formation  
377 and maintenance of the microphytobenthic biofilm, especially at the end of emersion time,  
378 through ion sequestration, negative charge, ECPS solubility, hydrophobicity [36].  
379 Unfortunately, statistic tests failed to demonstrate a significant effect of the emersion time on  
380 uronic acid dynamics.

381 Besides, the presence of inositol (8 – 15 %) was also interesting, given that no author has  
382 highlighted it. This molecule plays different and important roles as structural basis for a  
383 number of secondary messengers in eukaryotic cells and mostly as a major growth factor for

384 heterotrophic bacteria, implying that this sugar could be produced in the sediment, maybe by  
385 diatoms, to promote the symbiotic phenomenon led to the formation of benthic biofilm.

386 Bound ECPS, the active players involved in the biofilm construction

387 LMW bound fractions (Fig. 7) were obtained for a first time properly, without contamination  
388 by residual polymers since less glucose was found in these fractions, comparing to HMW  
389 fractions of Hanlon et al., (2006). Rhamnose (25 %), xylose (22 %), glucose (20 %) and  
390 galacturonic acid (13 %) were the main carbohydrates. HMW bound fractions were very  
391 similar to LMW and were mainly composed of rhamnose (30 %), xylose (28 %), galacturonic  
392 acid (18 %) and glucose (13 %). The Dowex-method allowed highlighting the presence of  
393 important levels of deoxy sugars in the fractions. Indeed, the percentage of rhamnose strongly  
394 increased during the emerged period, in contrast to the percentage of galacturonic acid, which  
395 decreased, and both xylose and glucose, which remained constant. Deoxy sugars, as  
396 rhamnose, can promote biostabilisation of sediments [15, 43] owing to their surface active  
397 properties. They can also influence the hydrophobic character of ECPS, by enhancing their  
398 adhesion to the sediment or by regulating salinity and desiccation phenomena through  
399 different retention rate of water at the surficial sediments [33]. Rhamnose is also a component  
400 of the outer cell membrane of certain bacteria and can be specifically recognized by lectins,  
401 transmitting signals to the cell- interior and conferring specific properties. Given some of  
402 these properties, the increase in the percentage of rhamnose in this fraction could be linked to  
403 the role of these bound ECPS, for example improving cell-cell interactions between bacteria  
404 and diatoms. Thus, bound carbohydrates could be closely involved into the formation of the  
405 extracellular matrix of the microphytobenthic biofilm and also into its biochemical short-term  
406 modifications in response to environmental conditions. Alternatively, rhamnose could play a  
407 role as a biochemical sensor to control “the biofilm entity”.

408 The amount of bacteria measured on the surface of the sediment seemed to provide another  
409 possible role of bound EPS. The results appeared to show a relationship between bacteria  
410 abundance and bound carbohydrate dynamics (Fig. 2B, Fig. 3B). The results suggested a  
411 potential negative effect of bound EPS on bacterial division, but no significant correlation was  
412 found between bacteria and bound carbohydrates.

413

#### 414 Residual fractions and C-storage

415 LMW and HMW residual fractions (Fig. 7) could correspond to intracellular and cell wall  
416 associated polymers but also consist of a mixture of different refractory EPS not totally  
417 extracted by the Dowex-method. Fractions could be compared to a part of HW and all HB  
418 fractions of Abdullahi et al. (2006) or Hanlon et al. (2006). The carbohydrate content was  
419 composed of glucose (18 to 43 %), galacturonic acid (19 %), mannose (18 %), xylose (10 %)  
420 and rhamnose (10 %). The observations of many authors indicated that, these fractions were  
421 very complex, due to the presence of proteins and sulfated groups, and the different  
422 monosaccharides detected could be involved in the formation of structural polymers,  
423 glycolipids or proteoglycans [24]. The important point to highlight was the slight decrease in  
424 the percentage of glucose during the first hours of emersion. However, towards the end of the  
425 emerged periods, the quantity of glucose doubled or tripled. This observation could be  
426 correlated to the capacity of diatoms “to sense the end of emersion”, by storing a maximum  
427 amount of carbon for their metabolism to survive during periods of darkness. This result was  
428 partly consistent with those obtained by previous authors who have shown that the relative  
429 abundance of glucose increased during a daytime emersion period [17]. Overall, it was  
430 considered that the majority of the glucosyl units were components of the storage polymers  
431 chrysolaminaran, composed of (1→ 3)  $\beta$ -glucan chains [40]. Concerning the other sugars

432 detected and their levels, the results were fairly close to those already observed in the  
433 literature.

#### 434 **Conclusion**

435 The aim of this work was to study the dynamics and biochemical compositions of EPS,  
436 produced on an intertidal mudflat, during the development of a microphytobenthic biofilm.  
437 This study highlighted the complexity involved in collecting and separating the different types  
438 of present EPS. The extraction procedure allowed focusing more accurately on the bound  
439 fractions, composed of specific carbohydrate monomers, *i.e.* large amounts of deoxy sugars  
440 and uronic acids. The changes in bound ECPS composition, especially through the levels of  
441 rhamnose, mannose or galacturonic acid, could play a major role during the development of  
442 the microphytobenthic biofilm by increasing binding forces and cell-cell interactions. The  
443 lack of fucose was a surprising observation and was correlated to the sampling period (winter)  
444 and the physiological state of the growing biofilm. Besides, the presence of inositol, detected  
445 for a first time *in situ*, was quite interesting. This sugar is commonly used for GC/MS analysis  
446 as an internal standard (*myo*-inositol) and this could explain why it was not identified as a  
447 component of ECPS. Significant amounts of this growing factor for bacteria were measured in  
448 the colloidal fractions, a fraction known as being a direct nutrient source for the heterotrophic  
449 bacteria constituting the microphytobenthic biofilm. In this way, the results showed that the  
450 colloidal fractions were not only used as a C-source but that they could have an impact on ion  
451 sequestration owing to high level of uronic acids. Finally, this study only focused on one  
452 sampling campaign during winter. It was shown that the greatest variations in sediment  
453 dynamics resulted from seasonal changes, modifying the concentration, type and structure of  
454 EPS accordingly (Underwood and Paterson, 2003). So, it should be interesting to check if  
455 these variations could be observed on this intertidal bay during summer.

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460 microbial biofilms in tidal flats” (contract ANR-06-BLAN-0393-01).

461

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577 formation of transparent exopolymer particles by bubble adsorption of seawater. *Limnol*  
578 *Oceanogr* 43:1860-1871
- 579

580 Table 1. Composition ( $\mu\text{g}/\mu\text{g chl } a$ ) in uronic acids, sulfates and proteins of the different  
 581 fractions extracted by the Dowex method from the Marennes-Oléron mudflat.

582

Fractions (LMW + HMW)	Colloidal EPS			Bound EPS			Residual polymers		
	Uronic acids	Sulfates	Proteins	Uronic acids	Sulfates	Proteins	Uronic acids	Sulfates	Proteins
2/18/08 - 1h	1.6 ± 0.9	0	0	1.3 ± 0.3	0	0	20 ± 5	8 ± 3	15 ± 1
2/18/08 - 2h	1.8 ± 0.3	0	0	1.4 ± 0.3	0	0	19 ± 7	8.0 ± 6	26 ± 6
2/18/08 - 3h	1.6 ± 0.9	0	0	1.4 ± 0.6	0	0	21.0 ± 6	9 ± 0.4	16 ± 0.7
2/18/08 - 4h	-	-	-	-	-	-	-	-	-
2/19/08 - 1h	1.2 ± 0.6	0	0	0.7 ± 0.3	0	0	28 ± 6	7 ± 2.0	21 ± 13
2/19/08 - 2h	1.1 ± 0.3	0	0	1.4 ± 0.8	0	0	19 ± 7	5 ± 3	18 ± 4
2/19/08 - 3h	1.5 ± 0.5	0	0	1.5 ± 0.6	0	0	25 ± 7	8 ± 1.0	20 ± 4
2/19/08 - 4h	-	-	-	-	-	-	-	-	-
2/20/08 - 1h	1.4 ± 0.5	0	0	1.3 ± 0.3	0	0	23 ± 9	9 ± 5	19 ± 4
2/20/08 - 2h	1.2 ± 0.3	0	0	0.9 ± 0.4	0	0	21 ± 15	11 ± 0.7	19 ± 8
2/20/08 - 3h	1.5 ± 0.4	0	0	0.8 ± 0.2	0	0	21 ± 9	10 ± 5	20 ± 11
2/20/08 - 4h	1.5 ± 0.3	0	0	0.9 ± 0.2	0	0	17 ± 5	12 ± 7	24 ± 8

583 ± : deviations were calculated from the heterogeneity of the different sampling squares and not from the true  
 584 replicates of the biochemical analyses (<5%)

585

586 Figure 1. Station where samples of surficial intertidal sediment were collected: Brouage  
587 mudflat.

588 Figure 2. Light climate (A) measured each minute throughout the sampling, cell density  
589 associated with Chl *a* in the first 10mm (B) and Chl *a* in the first 2mm (C) of the sediment.

590 Figure 3. Carbohydrate concentration ( $\mu\text{g glc equivs.}\mu\text{g chl } a^{-1}$ ) for colloidal (A), bound (B)  
591 and residual (C) fractions depending on the emerged period.

592 Figure 4. Distribution of LMW (% w/w) for colloidal (A), bound (B) and residual (C)  
593 fractions depending on the emerged period.

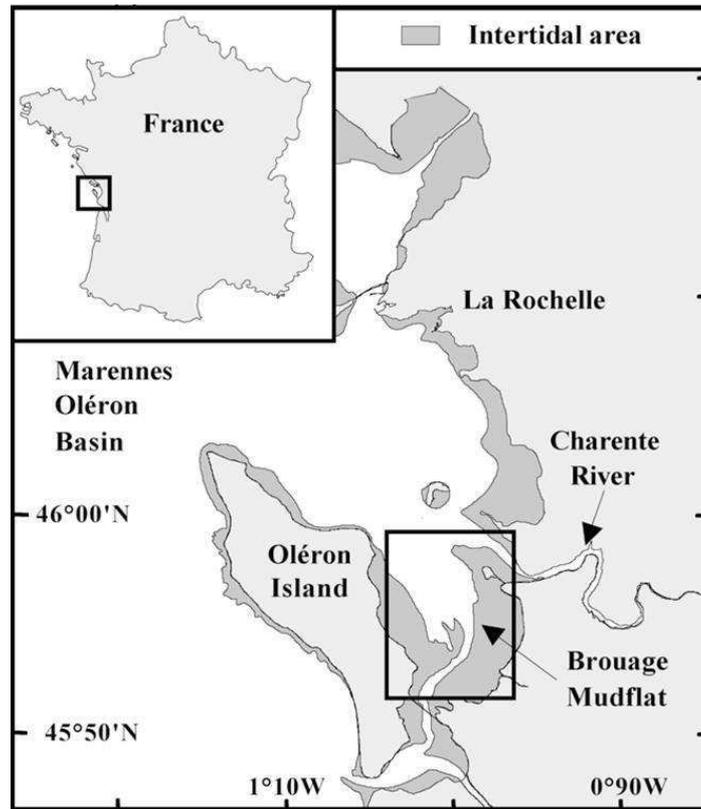
594 Figure 5. Monosaccharide composition (average for the 3 sampling sites) of the different  
595 fractions extracted from the mudflat. The variability within true sample replicates was less  
596 than 5%. Unknown: indicates the sum of traces which were not identified.

597 Figure 6. GC-MS chromatogram of carbohydrates detected in an HMW bound fraction.  
598 Ionization: Electronic Impact (EI, 70eV). Target ion: 40-650 m/z.

599 Figure 7. EPS types and location in the Marennes-Oléron mudflat, based on Underwood and  
600 Paterson's (2003) conceptual model for benthic diatom exopolymères. Composition in sugars  
601 (%) was averaged over the emersion period.

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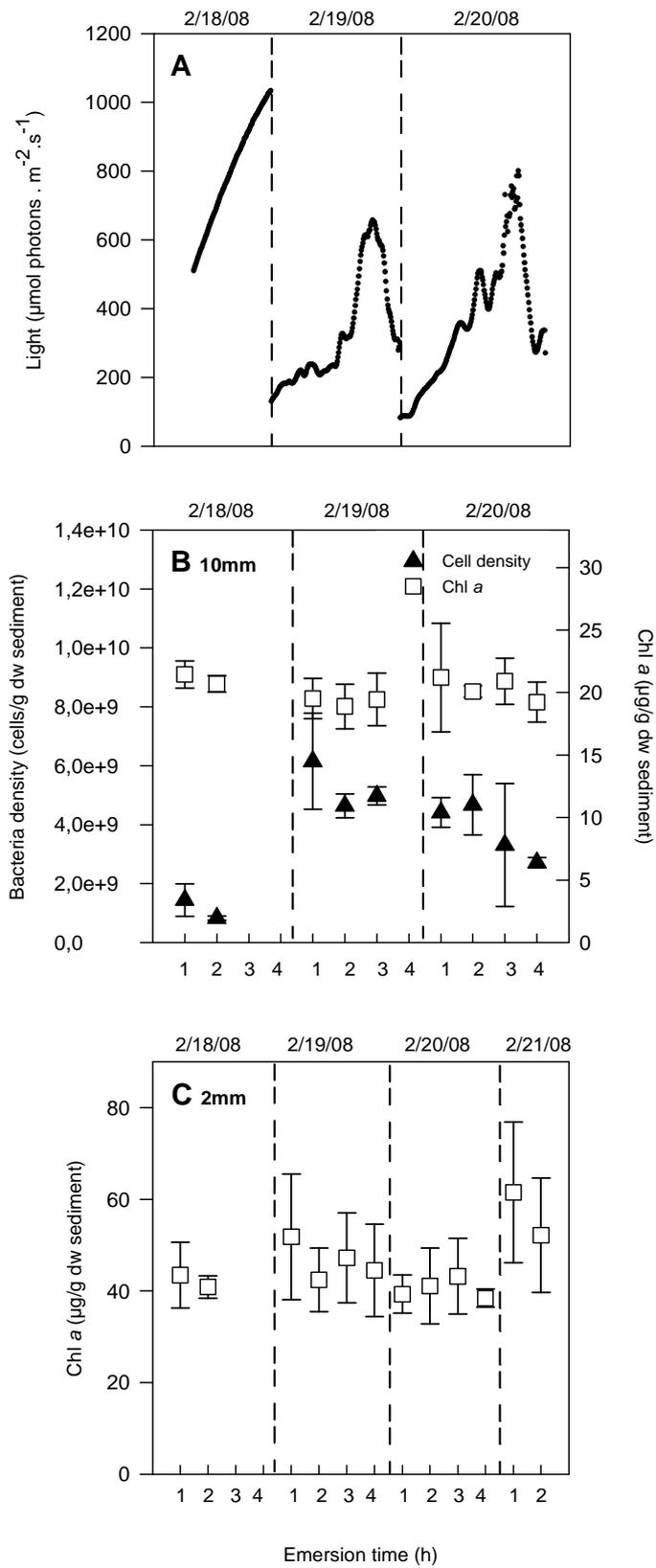
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FIGURE 1



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FIGURE 2

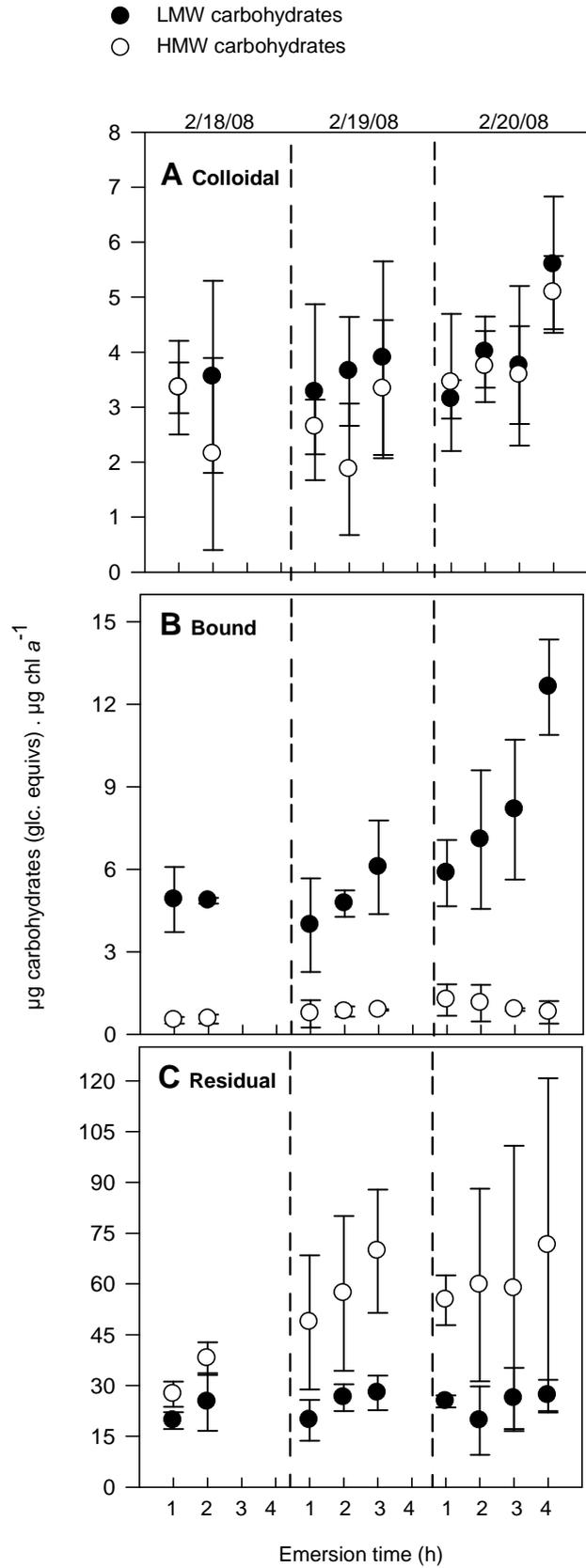
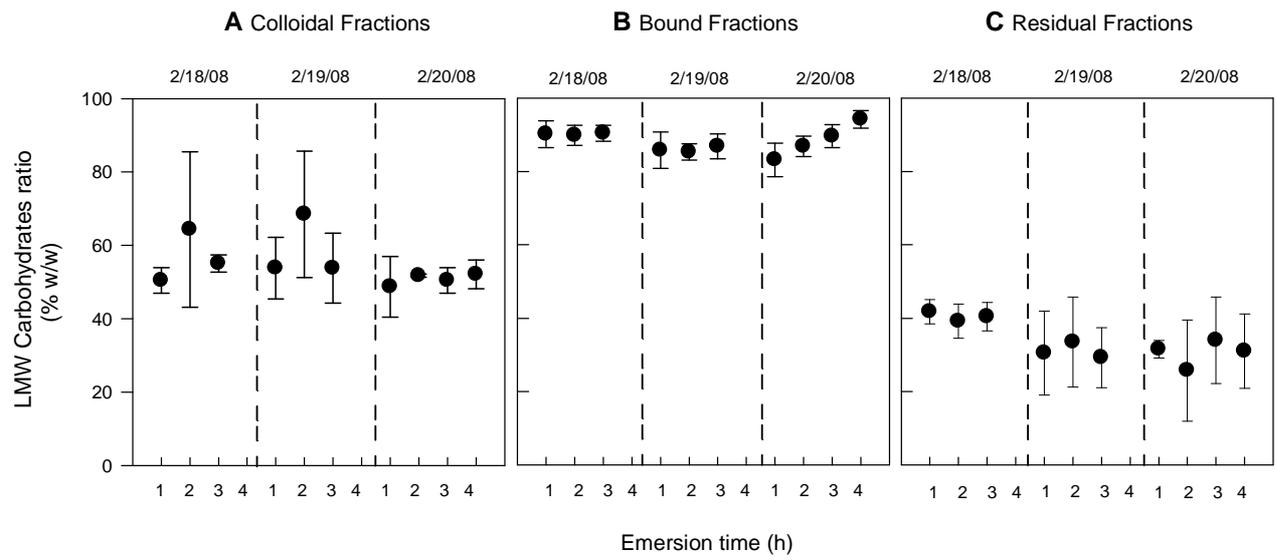


FIGURE 3



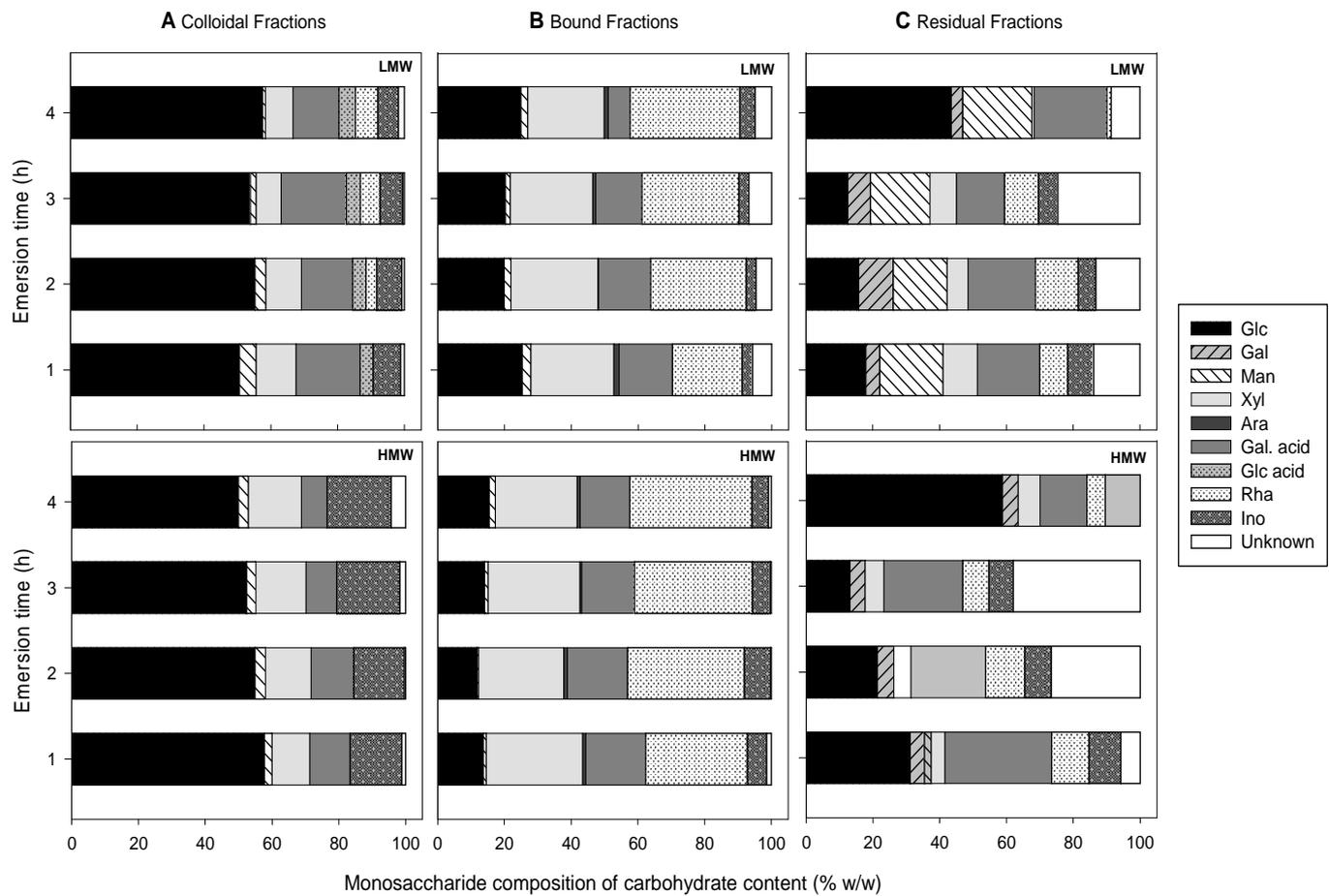
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FIGURE 4

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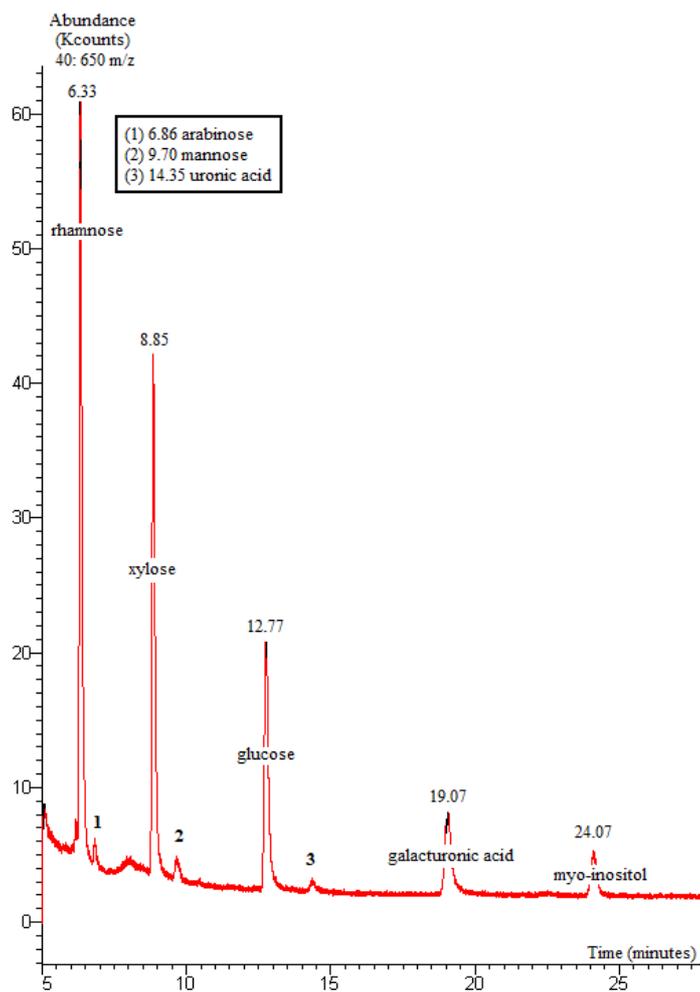


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FIGURE 5

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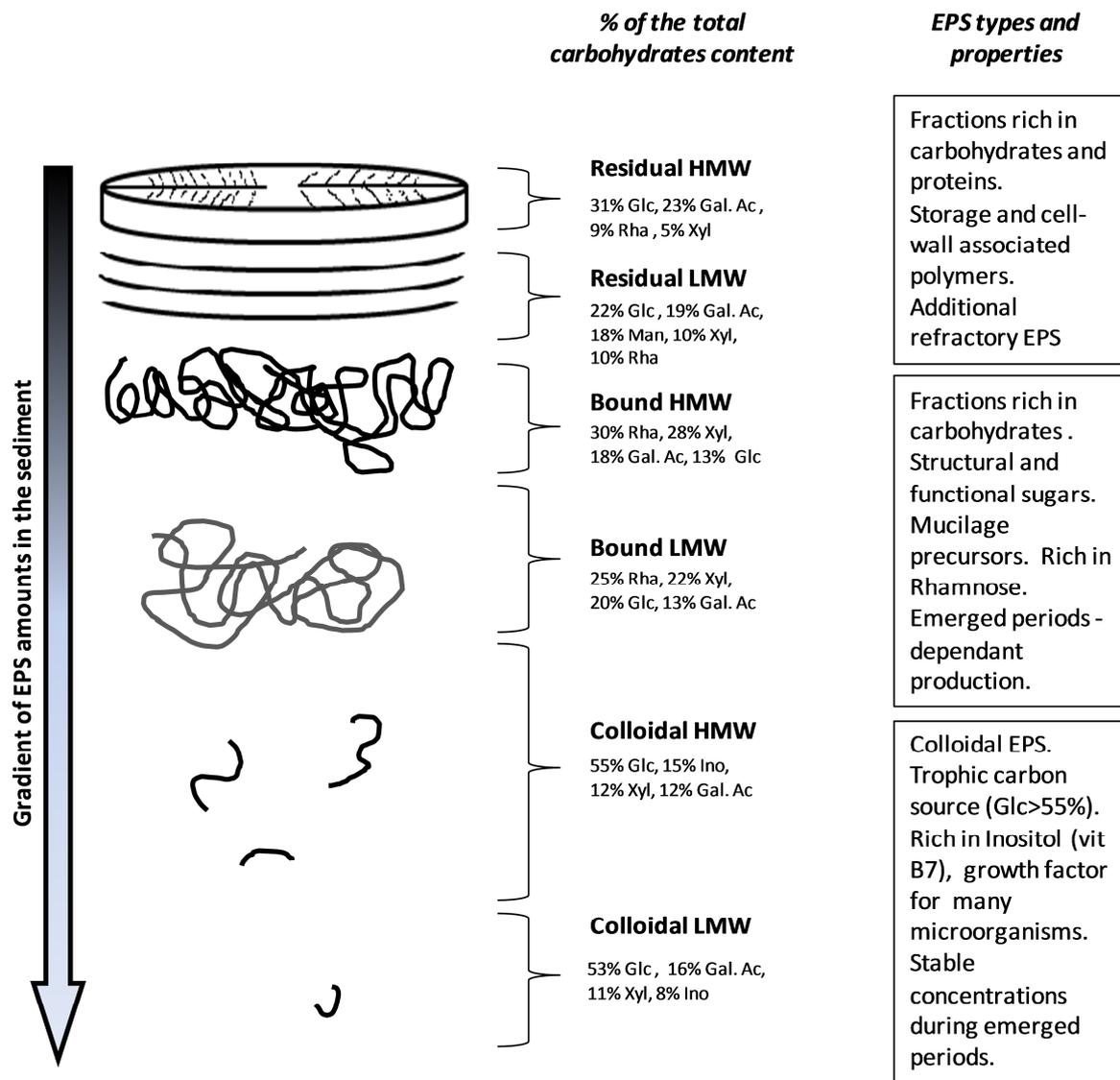
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FIGURE 6



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FIGURE 7