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1 **Disturbance increases microbial community diversity and**
2 **production in marine sediments**

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4 Pierre E. Galand^{1*}, Sabrina Lucas¹, Sonja K. Fagervold^{1†}, Erwan Peru¹, Audrey M. Pruski¹,
5 Gilles Vétion¹, Christine Dupuy², Katell Guizien¹

6
7
8 ¹Sorbonne Universités, UPMC Univ Paris 06, CNRS, Laboratoire d'Ecogéochimie des
9 Environnements Benthiques (LECOB), Observatoire Océanologique, F-66650, Banyuls sur
10 Mer, France

11 ²UMR 7266 Littoral, Environnement et Sociétés (LIENSs), CNRS – Université de La
12 Rochelle, Institut du littoral et de l'environnement, 17000 La Rochelle, France

13
14 [†]Current address: Sorbonne Universités, UPMC Univ Paris 06, CNRS, Laboratoire de
15 Biodiversité et Biotechnologies Microbiennes (LBBM), Observatoire Océanologique, F-
16 66650, Banyuls sur Mer, France

17
18 *Correspondence: Pierre Galand, pierre.galand@obs-banyuls.fr

25

26 **Abstract**

27 Disturbance strongly impacts patterns of community diversity, yet the shape of the diversity-
28 disturbance relationship remains a matter of debate. The topic has been of interest in
29 theoretical ecology for decades as it has practical implications for the understanding of
30 ecosystem services in nature. One of these processes is the remineralization of organic matter
31 by microorganisms in coastal marine sediments, which are periodically impacted by
32 disturbances across the sediment-water interface. Here we set up an experiment to test the
33 hypothesis that disturbance impacts microbial diversity and function during the anaerobic
34 degradation of organic matter in coastal sediments. We show that during the first 3 weeks of
35 the experiment, disturbance increased both microbial production, derived from the increase in
36 microbial abundance, and diversity of the active fraction of the community. Both community
37 diversity and phylogenetic diversity increased, which suggests that disturbance promoted the
38 cohabitation of ecologically different microorganisms. Metagenome analysis also showed that
39 disturbance increased the relative abundance of genes diagnostic of metabolism associated
40 with the sequential anaerobic degradation of organic matter. However, community
41 composition was not impacted in a systematic way and changed over time. In nature, we can
42 hypothesize that moderate storm disturbances, which impact coastal sediments, promote
43 diverse and productive communities. These events, rather than altering the decomposition of
44 organic matter, may increase the substrate turnover and, ultimately, remineralization rates.

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50 **Introduction**

51 One of the key features of microbial communities in most natural ecosystems is that they
52 harbor a tremendous diversity (Keller and Zengler, 2004). Community diversity is affected by
53 a number of biotic interactions but also by abiotic factors such the surrounding environmental
54 conditions. Environmental conditions change with time and can fluctuate dramatically when
55 impacted by episodic disturbances. A disturbance can be defined as “any relatively discrete
56 event in time that disrupts ecosystem, community, or population structure and changes
57 resources, substrate availability, or the physical environment” (Pickett and White, 1985). The
58 response of communities to disturbance has been a central interest in ecology studies for
59 decades. More specifically, investigations have aimed to resolve how disturbances in an
60 ecosystem influence the diversity of the communities (Sousa, 1984). In this respect, the
61 intermediate disturbance hypothesis (IDH) (Grime, 1973; Connell, 1978) suggests that the
62 relationship between diversity and disturbance can be represented graphically as a unimodal
63 distribution. Community diversity is low when disturbance levels are low and increases with
64 disturbance frequency or strength. If disturbance persists, however, diversity decreases again.
65 Nevertheless, recent studies have refuted the idea of a positive relationship between
66 disturbance and community diversity. A survey of empirical studies reported that non-
67 significant relationships were the most common (Mackey and Currie, 2001), and a recent
68 paper proposed, based on both empirical and theoretical grounds, that the IDH should be
69 abandoned (Fox, 2013). For microorganisms, soil experiments showed that diversity declined
70 with increasing disturbance frequencies, (Kim et al., 2013) and a negative diversity-
71 disturbance relationship was also observed in natural marine sediments (Boer et al., 2009).

72 Recently, it has been emphasized to incorporate genetic diversity into disturbance-
73 diversity studies because the topic remains a major knowledge gap (Banks et al., 2013).
74 Despite the recognition of the importance of genetic diversity in ecology (Hughes et al., 2008),

75 its role as a driver for microbial diversity and function remains poorly understood. Overall,
76 data on the IDH for microorganisms remains scarce because studies have mainly focused on
77 the impact of disturbance on community composition, rather than diversity (Allison and
78 Martiny, 2008; Berga et al., 2012; Yeo et al., 2013). Communities are hypothesized to be
79 resistant or resilient to disturbance when they are not definitely altered in their composition
80 (Allison and Martiny, 2008).

81 Episodic disturbances, which are thought to have an important role in maintaining
82 diversity, impact many of the ecosystems of the Earth. Coastal sediments are regularly
83 exposed to disturbances due to increases in turbulence that result from the dissipation of
84 energetic flows in the benthic boundary layer. These turbulences occur with different
85 strengths and at different temporal scales. Tidal flow generates periodic increases of
86 turbulence above the sediment with moderate intensity, whereas waves cause larger but more
87 sporadic turbulences, which lead to sediment resuspension. Turbulence intensity in the
88 benthic boundary layer controls diffusive fluxes across the sediment-water interface; as a
89 consequence, turbulence intensity fluctuations disturb the stratification of dissolved
90 compounds in the sediments and may affect organic matter degradation (Arzayus and Canuel,
91 2005).

92 Organic matter degradation is an essential service provided by heterotrophic
93 microorganisms (Ducklow, 2008). This is particularly true in estuarine sediments and deltaic
94 systems, where most of the terrestrial organic matter delivered by rivers is deposited before
95 being remineralized or buried (Hedges et al., 1997; Burdige, 2007). Understanding the fate of
96 terrestrial or marine organic matter exported down to sediments may help gain greater insight
97 into the cycling of carbon on a global scale and its possible alteration by climate change and
98 anthropogenic perturbations (Regnier et al., 2013; Reichstein et al., 2013). Degradation or
99 remineralization of the complex mixture of molecules composing sediment organic matter is

100 mediated by the combined activity of an array of both aerobic and anaerobic microorganisms
101 with different metabolic potential. Although aerobic processes are more effective, the anoxic
102 degradation rates are often similar to the oxic rates in surface sediments (Arndt et al., 2013).
103 The identity of the main players in the terminal degradation process and the structure of the
104 communities involved are not always well known. The composition of the microbial
105 communities can for instance be correlated to the origin and lability of organic matter
106 (Fagervold et al., 2014), the geochemical composition of the sediments (Jorgensen et al.,
107 2012) and pigment concentrations (Bienhold et al., 2012). The impact of disturbance, which
108 may disrupt the commonly observed vertical zonation of marine sediments, remains poorly
109 studied.

110 Here, we test the hypothesis that disturbance impacts microbial community diversity
111 in coastal sediments during organic matter mineralization. We conducted an experiment in
112 which sediment cores were regularly exposed to enhanced turbulence at the sediment-water
113 interface, without sediment resuspension, mimicking the effect of moderate swell events.
114 Sediments were enriched with plant-derived detritus to simulate the sedimentation of labile
115 terrestrial organic matter. Organic matter degradation was monitored during the course of the
116 experiment and so was the concentration of oxygen. Microbial abundance was measured as a
117 way to derive production. Bacterial and archaeal communities were described before and after
118 each disturbance event at two sediment depths by targeting both 16S rRNA genes and 16S
119 rRNA transcripts to identify the active fraction of the communities. We also constructed
120 metagenomes to test whether disturbance impacted potential sedimentary metabolic pathways
121 and to verify whether possible changes in community taxonomic composition were correlated
122 with changes in functional gene composition.

123

124

125 **Materials and methods**

126

127 **Experimental setup**

128

129 A total of 36 sediment cores enriched with duckweed (*Lemna sp.*) were placed in a
130 water tank incubated in the dark at 16 °C (see Supporting information for details). The
131 sediment cores were exposed to disturbance in the form of a spatially uniform diffusive
132 turbulence generated by an oscillating grid that controlled the diffusive fluxes across the
133 sediment-water interface (Fig. S1, (Lucas et al., 2015)). Turbulence frequency and strength
134 were chosen to simulate the disturbance generated by moderate swell events in the
135 infralittoral area. Overall, the experiment included four disturbance events preceded by calm-
136 periods. Twenty-four-hour disturbance events (turbulent velocity=6.15 cm s⁻¹) were separated
137 by calm-intervals of 10 days (turbulence < 1.18 cm s⁻¹). The entire experiment lasted for 5
138 weeks. The low turbulence in the calm-periods corresponded to turbulent conditions in the
139 benthic boundary layer during usual wind-driven circulation. The disturbance events did not
140 cause any sediment resuspension.

141 Sampling was conducted before and after each disturbance event. A total of 8 different
142 time points (T0-T7) were sampled (4 before disturbance and 4 after disturbance). For each
143 sampling, 3 cores were removed from the water tank. The cores were sliced and sediment
144 samples were taken from two layers: 0-5 mm (surface) and 5-10 mm (deep). Organic matter
145 concentrations and microbial abundances were measured on triplicate cores, and oxygen, fatty
146 acids and microbial molecular analysis were conducted on 1 of the 3 cores.

147

148

149

150 **Fatty acids, organic matter, oxygen and bacterial abundance**

151

152 Labile OM compounds, namely total carbohydrates, was assessed using colorimetric methods
153 according to previously published protocols (Dubois et al., 1956; Barnes and Blackstock,
154 1973; Bradford, 1976). Total fatty acids were extracted by direct acid transmethylation and
155 analyzed using a Saturn 2100T iontrap GC–MS instrument (VARIAN, Les Ulis, France)
156 equipped with a fused-silica-capillary column (Factor Four, VF-Waxms, 30mX0.25mm ID,
157 0.25 mm film thickness, VARIAN). Details of the extraction procedure and analytical
158 conditions are given in Bourgeois et al. (2011)(Bourgeois et al., 2011). Oxygen
159 concentrations were profiled at a 200 µm vertical spatial resolution every 15 min with a
160 Unisense O₂ Clark-type microsensor (Unisense, Aarhus, Denmark) from 20 mm above to 10
161 mm below the sediment-water interface. Time series of the average O₂ concentration in the
162 sediment layers (0-5 mm, 5-10 mm) were calculated from these profiles.

163 Prokaryotic abundance was measured by flow cytometry. Five milliliters of sediment
164 was fixed with 0.2 µm filtered formaldehyde solution (vol/vol, 2% final concentration), and
165 cells were separated from the sediment and homogenized according to the protocol described
166 by (Lavergne et al., 2014). One milliliter of sediment was diluted sequentially to 1/2000 with
167 0.01 M sodium pyrophosphate (NaPp) and was mixed by vortexing. Samples were then
168 incubated at +4 °C for 30 min before sonication (60 W for 30 s). Finally, an aliquot of the
169 sample was stained with SYBR Green I (1:10,000) for 15 min in the dark and was analyzed
170 by flow cytometry as previously described (Lavergne et al., 2014). Bacterial production was
171 derived from the increase in bacterial abundance and was expressed as the number of cells per
172 mL of sediment and per day. Production was calculated for each time point as:
173 $Production_{T_2} = (\ln(Ab_{T_2}) - \ln(Ab_{T_1})) / (T_2 - T_1)$, where Ab is the prokaryotic abundance and T is
174 the time.

175

176 **Nucleic acid extraction, RT-PCR and sequencing.**

177

178 DNA and RNA were co-extracted using the RNA PowerSoil Total RNA Isolation Kit and the
179 DNA Elution Accessory Kit (MoBio, Carlsbad, CA, USA) from 1g of frozen sediment
180 according to the manufacturer's protocol. RNA samples were DNase treated using the
181 RNase-Free DNase Set (Qiagen, Valencia, CA, USA) and concentrated with the RNeasy
182 MinElute Cleanup Kit (Qiagen). The RNA were reverse-transcribed with random primers
183 using the SuperScript III Reverse Transcriptase kit (Invitrogen, Carlsbad, CA, USA) and
184 tested for the presence of contaminating genomic DNA by performing PCR before the reverse
185 transcription.

186 Portions of the 16S rRNA genes were amplified with the bacterial primer pair 27F (5'-
187 AGRGTTTGATCMTGGCTCAG-3') and 519R (5'-GTNTTACNGCGGCKGCTG-3'), and
188 archaeal primers 349F (5'-GYGCASCAGKCGMGAAW-3') and 806r (5' -
189 GGACTACVSGGGTATCTAAT-3'). A 30 cycle PCR with the HotStarTaq Plus Master Mix
190 Kit (Qiagen, Valencia, CA) was conducted under the following conditions: 94 °C for 3
191 minutes, followed by 28 cycles of 94 °C for 30 seconds, 53 °C (bacteria) or 50 °C (archaea) for
192 40 seconds and 72 °C for 1 minute, after which a final elongation step at 72 °C for 5 minutes
193 was performed. Pyrosequencing was conducted by a commercial laboratory (Research and
194 Testing Laboratory, Lubbock, TX, USA) on a Roche 454 FLX (Brandford, CT, USA) using
195 commercially prepared Titanium reagents. Sequences have been deposited to the GenBank
196 Sequence Read archive under number SAMN04126915- SAMN04126946.

197 For the metagenomes, libraries for shotgun sequencing were prepared using Nextera
198 DNA Sample Preparation Kit (Illumina, San Diego, CA, USA) with 50 ng of DNA from each
199 sample. Library insert size was determined by Experion Automated Electrophoresis Station

200 (Bio-Rad, Hercules, CA, USA). The insert size of the libraries ranged from 300 to 850 bp
201 (average 500bp). Pooled libraries (12pM) were loaded to a 600 Cycles v3 Reagent cartridge
202 (Illumina) and the sequencing was performed on Miseq (Illumina) by a commercial laboratory
203 (Research and Testing Laboratory, Lubbock, TX, USA). Metagenomes are archived in MG-
204 RAST under accession numbers 4612993.3, 4612994.3, 4612995.3, 4612996.3 and
205 4612997.3.

206

207 **Sequence analysis**

208

209 All reads that had a mismatch with the 16S rRNA primers, contained ambiguous nucleotides
210 (N) or were longer than 390 after the forward primer, were removed. Then, reads that had \geq
211 3% of bases with Phred values < 27 (0.2% per-base error probability) were removed. These
212 steps are recommended to ensure that when clustering at 97%, the influence of erroneous
213 reads is minimized (Huse et al., 2010). Sequences were clustered at a 97% threshold using
214 Uclust (Edgar, 2010). Representative sequences of each OTU were classified by comparison
215 with Blast to the Greengenes database (DeSantis et al., 2006). Sequence analyses were
216 conducted with Pyrotagger (Kunin and Hugenholtz, 2010). To compare bacterial communities
217 for diversity analysis, all samples were randomly re-sampled to the size of the sample
218 containing the fewest sequences (n=950 for Bacteria and n=424 for Archaea) using Daisy
219 Chopper (Gilbert et al., 2009).

220 For metagenomes, sequence ends were cut to remove adapters. Sequences were
221 considered low quality when >5 bases had a phred score <15 and were subsequently removed.
222 FastqJoin was used to merge paired end reads when possible with a minimum overlap setting
223 of 8 bp and a maximum difference of 10%. Both paired and unpaired sequences were kept for
224 automated annotation in MG-RAST using Hierarchical Classification subsystems against the

225 KEGG Orthology database. Annotations were conducted with a maximum e-value cutoff of
226 $1e^{-10}$, a minimum 60% identity cutoff and 15 amino acids as the minimum alignment length
227 cutoff. Identified microbial gene families (specified by KEGG orthology groups) were
228 grouped into metabolic pathways (Table S1). Metagenome reads were normalized by evenly
229 resampling at random to 1,254,500 annotated reads per sample to allow comparison.

230

231 **Data analysis**

232

233 Bacterial diversity was estimated for both the RNA and DNA fractions by calculating the
234 Shannon diversity index (H') and by the standardized effect size (SES) (Kembel et al., 2010),
235 which is a standardized measure of phylogenetic diversity. Phylogenetic diversity was
236 computed using the Picante package (Kembel et al., 2010) in R. To calculate SES, 300-bp-
237 long representative sequences for each OTU were aligned using MUSCLE (Edgar, 2004) and
238 the alignment was then cleaned to remove nonoverlapping sequence regions. A phylogenetic
239 tree was constructed using FASTTREE (Price et al., 2010). The observed phylogenetic
240 diversity was compared to the average phylogenetic diversity in a randomly generated
241 community (null model) and divided by the standard deviation of phylogenetic distances in
242 the null model (Webb et al., 2008). The null model randomizes community data matrix with
243 the independent swap algorithm maintaining species occurrence frequency and sample species
244 richness (Kembel, 2009). SES is equivalent to -1 times the Nearest Relative Index (NRI)
245 (Webb et al., 2002). Faith's PD (Faith, 1992) is the most common measure of phylogenetic
246 diversity, but because the number of taxa in a sample affects PD and because the number of
247 taxa varied significantly between our samples, we chose to use SES instead. Positive SES
248 values indicate greater phylogenetic distance among co-occurring species than expected by
249 chance, whereas negative values indicate small phylogenetic distance.

250 The Bray Curtis similarity index was computed to compare the community and functional
251 gene composition between samples and to conduct cluster analysis. The effects of disturbance
252 and time on community composition were tested with a permutational multivariate analysis of
253 variance using distance matrices as implemented in the adonis function in the Vegan package
254 (v.2.3-0) in the R software (v.3.1.2). To identify OTUs that had a significant difference in
255 abundance between groups we used an ANOVA with a Tukey-Kramer post-hoc test as
256 implemented in STAMP (Parks et al., 2014).

257

258 **Results**

259 **Dynamics of oxygen, carbohydrates and fatty acid concentrations**

260 Each of the four disturbance events, which lasted for 24 h (T0-T1, T2-T3, T4-T5, and T6-T7),
261 oxygenated the water above the sediment, resulting in O₂ saturation. Oxygen concentrations
262 in the overlying water decreased at the beginning of each interleaved calm-interval (T1-T2,
263 T3-T4, and T5-T6) (Fig. 1a). Oxygen concentrations in the sediment did not follow the same
264 trend as in water: the concentration increased after the first disturbance and remained low at
265 0.20 mg L⁻¹ on average in the upper layer of the sediments during the following calm interval
266 (T1-T2) (Fig. 1a). After the second disturbance event (T2-T3), the oxygen concentrations in
267 the upper part of the sediment increased with a peak of 0.60 mg L⁻¹ three days after the end of
268 the disturbance, and then decreased to 0.40 mg L⁻¹ at the end of the calm interval (T3-T4) (Fig.
269 1a). The third disturbance (T4-T5) immediately increased oxygen concentrations up to 1.50
270 mg L⁻¹ in the upper sediment layer, but the concentrations rapidly dropped to lower levels
271 than during the first and second calm intervals (Fig. 1). The last disturbance (T6-T7)
272 increased the oxygen concentration in the sediment to 0.50 mg L⁻¹.

273 In summary, turbulent disturbance oxygenated the mesocosm water, thereby
274 increasing oxygen flux to the sediment. However, the average concentrations in the sediment

275 varied during the course of the experiment, and the 0-5 mm surficial sedimentary layer
276 remained anoxic on average ($<0.6 \text{ mg L}^{-1}$), with the exception of the first hours after the T4-
277 T5 disturbance, during which the sediment was in hypoxia. The deeper part of the sediment
278 (5-10 mm) was always anoxic (data not shown).

279 The average carbohydrate concentration was $8.1 \mu\text{g g}^{-1}$ in the surface sediment layer at
280 the beginning of the experiment and decreased to $5.4 \mu\text{g g}^{-1}$ at T2 (Fig. 1b). The
281 concentrations remained low, with values varying from 4.5 to $5.6 \mu\text{g g}^{-1}$ of dry sediment.
282 Among the fatty acids quantified by GC-MS, four were characteristic of the fatty acid
283 signature of the duckweed used to enriched the sediments (C16:0, C18:1 ω 9 cis, C18:2 ω 6 cis
284 and C18:3 ω 3) and accounted for 77% of the fatty acids in the sediments at T0. The
285 concentrations of these biomarkers decreased strongly from T0 to T3 and remained at low
286 concentrations after that (Fig. 1b), which indicates that the decrease in carbohydrate
287 concentrations reflected the degradation of duckweed detritus in the surface sediment layers.

288

289 **Prokaryote production and community diversity**

290 Bacterial community diversity, represented by the Shannon index (H), varied during the
291 course of the experiment in both the surface and deep layers (Fig. 2a) and for both the RNA
292 and DNA fractions (Fig. S2). For the RNA fraction, the surface and deep sediment bacterial
293 communities had similar patterns (Fig. 2a). Bacterial community diversity increased sharply
294 after each of the 3 first disturbance events and remained stable or even decreased during calm
295 periods (Fig. 2a). For the DNA fraction, the surface and deep layers had similar diversity
296 dynamics patterns, with the exception of the T2-T3 disturbance event (Fig. S2), but the
297 patterns differed from the RNA fraction. The first disturbance resulted in a sharp decrease in
298 bacterial diversity (Fig. S2). Then, increases were observed with turbulence, and there was a
299 decrease at the end of the experiment.

300 Similar to community diversity, the phylogenetic diversity for the RNA fraction,
301 measured as SES, increased in surface sediments after each of the first three disturbance
302 events (Fig. 2b) and decreased after T5. Moreover, the SES values were negative at T0, T1
303 and T2 for both sediment depths (Fig. 2b), positive in the T3 to T5 samples, and then negative
304 again in the T6 and T7 samples. The deep sediment SES patterns were similar to the surface
305 patterns. All SES values were significant against a null model, with the exception of the
306 surface sample at T1.

307 In surface sediments (0-5 mm), the prokaryote production, expressed as the number of
308 cells per mL of sediment per day, increased after each disturbance event and decreased during
309 calm intervals (Fig. 2c). In deep sediments (5-10 mm), we observed an opposite trend, with a
310 decrease in prokaryote production after each turbulence events (Fig. 2c).

311

312 **Effect of time and disturbance on community composition**

313 We calculated the Bray Curtis similarity index between each possible sample pair to compare
314 community composition similarity with time. In both surface and deep sediment samples, the
315 Bray Curtis values were higher when samples were separated by shorter time intervals (Fig.
316 3a), and the similarity decreased as the time separating communities increased (ANOVA,
317 $F_{1,26}=22.76$, $p<0.001$), which indicated that turbulence (samples separated by 1 day) had less
318 effect on community composition compared to time.

319 We further tested the effect of disturbance and time with a permutational multivariate
320 analysis of variance using distance matrices (adonis statistics). Disturbance did not impact the
321 community composition at the RNA level ($F_{1,8}=0.04$, $p= 0.73$), whereas time was a significant
322 factor impacting the community composition ($F=0.36$, $p= 0.001$). There was no difference in
323 community composition between depths ($F=0.046$, $p= 0.65$).

324

325 **Community succession**

326 The composition of microbial communities was compared between samples with a
327 hierarchical cluster analysis based on the Bray Curtis index. Active bacteria communities
328 (RNA) were separated into 3 main clusters (Fig. 4a). One contained samples taken at T0, T1
329 and T2, one contained samples taken at T3, T4 and T5 and the last contained samples from T6
330 and T7. At T0, T1, T2, T6 and T7, surface and deep sediment communities grouped together,
331 showing similar community composition between depths. At T3 and T5, the deep and surface
332 communities were separated, suggesting a stronger stratification of the communities with
333 depth (Fig. 4a). Pre- and post-disturbance samples grouped together, indicating that
334 disturbance events had a lower impact on the composition of the active communities than
335 calm periods, despite the increase of community diversity. The second disturbance event
336 represented an exception because it induced a major change in community composition (Fig.
337 4a). The T5 and T6 communities were separated in two different clusters, which indicated that
338 the third calm interval had a strong impact on the active bacterial community composition.

339 At the phylum/class level, the first cluster of active bacteria (RNA fraction) (T0, T1,
340 and T2) contained a majority of *Gammaproteobacteria*, *Fusobacteria* and *Firmicutes*
341 sequences (Fig. 5). In the second cluster (T3, T4, and T5), the proportion of
342 *Deltaproteobacteria* and *Bacteroidetes* sequences increased. The third cluster (T6 and T7)
343 was composed mostly of *Firmicutes* (> 70% of the sequences). The composition of the
344 bacterial communities described with DNA was similar to that observed at the RNA level, but
345 there were more *Bacteroidetes* sequences and fewer *Deltaproteobacteria*.

346 At the OTU level, the first cluster (T0, T1, and T2) had a higher abundance of
347 sequences belonging to OTU 1 and OTU 1657 (Fig. 4a, Fig. S3), which were similar to
348 sequences from *Propionigenium maris* (*Fusobacteria*) (98% similarity), an anaerobic strain
349 found in sediments that can ferment glucose and succinate primarily to propionate (Watson et

350 al., 2000). Other specific OTUs included OTU 80, 98% similar to the sediment sequences
351 belonging to the order *Clostridiales* (*Firmicutes*), and OTU 27, identified as *Vibrio kanaloae*
352 (*Gammaproteobacteria*) (99% similarity), which can ferment different compounds as the sole
353 carbon source (Thompson et al., 2003). The second cluster (T3, T4, and T5) had more
354 sequences belonging to OTU 239, OTU 943, OTU 137 and OTU 84 (Fig. 4a, Fig. S3). All
355 belonged to the order *Desulfobacterales* (*Deltaproteobacteria*), a group of sulfate-reducing
356 bacteria. The third cluster (T6 and T7) was characterized by the presence of OTU 476, OTU
357 302, OTU 8 and OTU 2, all belonging to *Clostridiales* (*Firmicutes*), an order with many
358 fermenting bacteria.

359 For Archaea, active communities (RNA fraction) were separated in 2 main clusters
360 (Fig. 4b). The first contained samples from T0 and T1, and the second cluster grouped the
361 other samples, with the exception of the deep T2 sample. At the phylum/class level, the T0-T1
362 cluster contained mostly *Thaumarchaeota* (>85% of the sequences), whereas the second
363 cluster (T2-T6) contained mostly *Methanomicrobia* sequences (Fig. S4). At the DNA level,
364 the *Methanomicrobia* were less abundant, and there were more sequences from the
365 SAGMEG-1 and C2 group, as well as *Methanobacteria*.

366 At the OTU level, the T0-T1 cluster had a higher abundance of sequences belonging to
367 OTU 54, OTU 3 and OTU 4 (Fig. S5), which were 99%, 98% and 97% similar to the
368 *Candidatus Nitrosopumilus koreensis* AR1 (*Thaumarchaeota*), respectively (Park et al., 2012).
369 The second cluster (T2-T6) was characterized by OTU 8, which was 99% similar to the
370 methanogen *Methanococcoides methylutens* (*Methanomicrobia*), and OTU 13 and OTU 711,
371 which were 99% and 97% similar, respectively, to *Methanolobus oregonensis*
372 (*Methanomicrobia*). All belong to the family Methanosarcinaceae, a group known for
373 performing all pathways of methanogenesis.

374

375 **Functional genes dynamics**

376 We constructed metagenomes for surface sediment samples T2, T3, T4, T5 and T6 and
377 obtained a total of ca. 66.8 million sequences after quality check (Table S2). The Bray-Curtis
378 index, calculated from the abundance of annotated sequences found in the functional genes
379 categories, showed that there was no correlation between the time separating two samples and
380 the functional gene composition of the samples (ANOVA, $F=0.03$, $p=0.85$) (Fig. 3b).

381 Within the annotated metagenome sequences, we identified the level 2 gene categories
382 that varied the most between samples (Fig. S6). The number of sequences associated with
383 membrane transport was highest at T2 and then decreased to similar lower levels. Energy
384 metabolisms showed an opposite trend. There were fewer sequences at T2, and after an
385 increase at T3, the number of sequences did not change significantly. Sequences for genes
386 involved in cell motility were abundant in samples T2 and T4 and then decreased at T5 and
387 again at T6. Cell growth and death increased after each turbulence event.

388 We then specifically targeted some metabolic pathways present in sediments that may
389 be impacted by disturbances. Genes typical for methanogenesis, sulfate reduction, nitrate
390 reduction, aerobic respiration, fermentation and nitrification were identified (Table S1). The
391 genes that had the highest number of sequences were chosen to monitor the pathway
392 dynamics in the sediments (Table S1). All of the gene sequences increased in abundance after
393 the first turbulence event, with the exception of genes involved in fermentation (Fig. 6). After
394 the second turbulence event, genes for sulfate reduction and aerobic respiration showed large
395 increases (Fig. 6a), whereas genes for nitrate reduction and nitrification showed small
396 increases (Fig. 6b). The number of sequences associated with sulfate reduction and aerobic
397 respiration increased overall over time during the course of the experiment, whereas the
398 number of sequences for genes associated with methanogenesis, nitrification and nitrate
399 reduction decreased after the first turbulence event (Fig. 6).

400 To compare the metagenome and amplicon results, we counted the number of 16S
401 rRNA amplicon sequences identified as methanogens (belonging to the classes
402 *Methanobacteria*, *Methanococci* and *Methanomicrobia*) and sulfate reducers (order
403 *Desulfobacterales*). For methanogens, the DNA and RNA counts showed similar patterns,
404 which indicated that the microbes that were present were also active. The disturbance events
405 resulted in an increase of the number of methanogen 16S rRNA sequences. For sulfate
406 reducers, the DNA and RNA patterns were also similar (Fig. S7). The second and third
407 disturbance events resulted in an increase in the number of sequences in a pattern similar to
408 that observed for metagenomic functional genes (Fig. 6).

409

410 **Discussion**

411 We showed that disturbance increased the diversity of marine sediment bacterial communities
412 during the first three weeks of incubation in a mesocosm experiment. Our finding can be
413 interpreted in the context of the intermediate disturbance hypothesis, IDH (Grime, 1973;
414 Connell, 1978), which predicts that disturbance will increase community diversity up to a
415 certain level of disturbance strength or frequency, after which diversity will decrease. The
416 increase in diversity is hypothesized to originate from the creation of niches because
417 disturbances contribute to environmental heterogeneity. These niches are colonized by new
418 species and the effect of competitive dominance is reduced (Violle et al., 2010). If disturbance
419 is too frequent, extinction will occur, only the few species able to cope with an unstable
420 environment will grow, and diversity will decrease. In our experiment, diversity increased
421 until T5 (3 weeks) and then started to decrease. The last disturbance event did not augment
422 diversity, and we propose that we observed the decrease predicted by the intermediate
423 disturbance hypothesis. The IDH has been challenged recently, however, and the shape of the
424 diversity-disturbance relationship remains a matter of discussion (Mackey and Currie, 2001;

425 Randall Hughes et al., 2007; Fox, 2013). This is the first report of a positive disturbance –
426 diversity relationship for benthic microorganisms. The hypothesis was tested previously with
427 *in situ* data from marine sediment, where depth was used as a proxy for disturbance (Boer et
428 al., 2009), but only a weak relationship was observed. A number of studies have been
429 conducted on soil microorganisms, in which no relationship (Fierer et al., 2003; Wakelin et al.,
430 2010; Deng et al., 2011) or only a negative impact of disturbance has been reported (Kim et
431 al., 2013). Here, the positive relationship was observed both at the beginning of the
432 experiment, when the nutrient load was high, and later, when the organic carbohydrate
433 concentrations were significantly lower. This is an important point because community
434 diversity may be directly influenced by the resources available in the ecosystem. It has been
435 shown, for instance, that high nutrient availability may reduce the number of species (Torsvik
436 et al., 2002) and that the decline in energy over time may impact the level of diversity
437 (Scholes et al., 2005). Our observation of diversity increasing independent of nutrient
438 availability argues for a direct effect of disturbance on diversity. It should, however, be noted
439 that the availability of electron acceptors is also very important in determining sediment
440 community composition. The differences in cell numbers observed between surface and deep
441 layer may reflect differences in oxygen availability between depths.

442 Interestingly, phylogenetic diversity was also positively impacted by disturbance,
443 which indicates that disturbance promoted the co-occurrence of phylogenetically distinct
444 species. Bacteria with distant common ancestors are more likely to be ecologically different.
445 Thus, the presence of distantly related species after disturbance supports the notion that
446 diversity increased because additional niches were created and occupied by ecologically
447 different species, which reduced competitive exclusion. Inversely, the low values measured
448 toward the end of the experiment indicate phylogenetic clustering, which can be interpreted as
449 communities structured by environmental filtering (Webb et al., 2002). The environment

450 selects a subset of ecologically similar taxa able to dominate under the given environmental
451 conditions.

452 We also showed that in surface sediments, diversity bursts were accompanied by an
453 increase in prokaryote production. The finding of stimulated production is supported by the
454 observation of a relative increase, after each disturbance event, of genes that belonged to the
455 “cell growth and death” category, which may indicate that cells were actively growing. The
456 link between production and diversity is a key topic in ecology (Naeem et al., 2009; Cardinale
457 et al., 2012) and a positive relationship is usually observed for community diversity (Gillman
458 and Wright, 2006; Cardinale et al., 2012). For marine microbes, contrasting results show a
459 negative relationship for bacterial richness (Reinthal et al., 2005; Obernosterer et al., 2010)
460 but a positive relationship between production and the phylogenetic diversity of active
461 bacteria (Galand et al., 2015). High-diversity communities may use resources more efficiently
462 through positive interactions and/or niche partitioning (complementary effect); alternatively,
463 more diverse communities are more likely to contain highly productive organisms (sampling
464 effect) (Loreau and Hector, 2001). In our experiment, the relationship did not hold in the
465 deeper layer of the sediments, where production decreased with disturbance. Negative
466 relationships between production and diversity have been shown for bacteria under controlled
467 experimental settings (Horn et al., 2003; Becker et al., 2012), suggesting that other factors
468 may constrain the relationship. We also found that prokaryote production decreased during
469 calm intervals. Whereas increased microbial abundance has been detected at intermediate
470 disturbance level in soil (Kim et al., 2013), our data suggest that high production was not
471 maintained after disturbance events, or that only a few of the sediment species could maintain
472 growth as time passed.

473 We identified specific groups of active microorganisms that increased in relative gene
474 abundance when the microbial biomass increased. Methanogens and sulfate reducers both

475 increased in sequence abundance with disturbance and may be contributors to the overall
476 biomass bursts. The question remains as to why these groups were stimulated and others, such
477 as fermenters, were inhibited. We can speculate that disturbance made fermentation end
478 products more available and that they were then used by methanogens and sulfate reducers,
479 which both represent the final step of anaerobic organic matter remineralization.

480 Although we observed changes in sequence abundance for some groups, our
481 experiment showed that disturbance did not have a systematic impact on the composition of
482 active communities, which indicates that the communities overall changed according to the
483 time of incubation. The presence of a temporally dynamic community implies that
484 communities will not recover to their initial composition after disturbance. In such a dynamic
485 ecosystem, resilience can probably not be observed. Our results suggest that for defining a
486 general response to disturbance in terms of community composition (Allison and Martiny,
487 2008) an ecosystem has to be stable or in a steady state. However, stability, which results
488 from a combination of biotic and abiotic factors, is never or rarely observed in coastal
489 sediments impacted by seasonal variations in physical forcing and nutrient loads.

490 We identified three main stages during our incubation that corresponded to different
491 community compositions. The first period was dominated by sequences affiliated with
492 potential fermenting bacteria and with archaea identified as ammonia oxidizers. During the
493 second stage, we detected more sequences belonging to sulfate reducing bacteria and
494 methanogenic archaea. For the last stage, methanogens were still present, and sequences
495 associated with potential fermenters, different from the first ones, appeared. We hypothesize
496 that the general pattern of changing communities over time followed the transformation of
497 organic matter contained in the sediments.

498 The overall potential microbial metabolism did not change as much as the composition
499 of the communities during the course of the incubation. The first stage was the most

500 dissimilar, with relatively fewer genes associated with energy metabolism but more
501 membrane transport, whereas cell motility decreased toward the end of the incubation.
502 However, the comparison of the gene function composition did not show significant changes
503 with time, which indicated that there was a certain level of functional redundancy within the
504 communities. Assemblies that appear different at the 16S rRNA gene level may actually
505 contain similar metabolic functions. These functions would be maintained during the course
506 of the experiment, but the identity of the microorganisms bearing these functions would
507 change with the evolving environmental conditions. More studies are needed to resolve the
508 question of microbial functional redundancy because our observation remains limited to one
509 type of experimental microbial ecosystem.

510

511 **Conclusion**

512 We showed that disturbance had a positive effect on sediment microbial communities by
513 increasing their production and both their community and phylogenetic diversity during the
514 first 3 weeks of the experiment. This result implies that disturbance stimulated the overall
515 community growth and promoted the development of many phylogenetically different
516 microorganisms. These diverse communities occupy a wide range of niches and consume a
517 variety of substrates produced during the sequential anaerobic degradation of organic matter.
518 In nature, we can hypothesize that the moderate storm disturbances that regularly impact
519 coastal sediments also promote diverse communities. These events, rather than disturbing the
520 decomposition of organic matter, probably increase the substrate turnover and, ultimately,
521 remineralization rates.

522

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527

528 **Author Contributions:**

529 SL, KG designed the experiments, PEG, SL, AP, EP, SF, GV, CD, KG carried out the work, SL,

530 PEG, KG interpreted the results, PEG wrote the manuscript that was revised and

531 improved by all the coauthors

532

533 **Conflict of Interest Statement:**

534 The authors declare that the research was conducted in the absence of any commercial

535 or financial relationships that could be construed as a potential conflict of interest.

536

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706 **Fig. 1.** Oxygen concentrations in the water (crosses) and sediments (triangles) along the
707 course of the experiment (a), and Lemna sp. fatty acids (open circles) and total
708 carbohydrate concentrations (filled circles) in the experimental sediments (b). Lemna sp.
709 fatty acids are represented by the sum of four characteristic saturated,
710 monounsaturated and polyunsaturated fatty acids (see material and methods). Grey
711 bars represent the disturbance events.

712
713 **Fig. 2.** Changes in the RNA-based community diversity (a) and phylogenetic diversity (b),
714 and prokaryote production expressed as the change in the number of cells per mL of
715 sediment and per day (c) in the surface and deep sediment layers along the course of the
716 experiment. Grey bars represent the disturbance events.

717
718 **Fig. 3.** Differences in the taxonomic community composition between samples (a) and
719 differences in the functional community composition (b) in relation to the time
720 separating the samples. Differences in the community composition are calculated with
721 the Bray-Curtis similarity index based on 16S rRNA sequences for the taxonomic
722 comparison (a) and are based on the functional genes from the metagenomes annotated
723 against the KEGG orthology database (b).

724
725 **Fig. 4.** Hierarchical clustering of the composition of the bacterial (a) and archaeal (b)
726 communities based on 16S rRNA transcripts sampled along the course of the experiment
727 (T0-T7) from the surface (s) and deep (d) layers of the sediment. The heatmap shows
728 the relative abundance of each operational taxonomic unit (OTU) (rows). The names of
729 the bacterial OTUs that were found to characterize each of the three main clusters of
730 samples (column) are indicated.

731

732 **Fig. 5.** Taxonomic affiliation and proportion of the bacterial 16S rRNA sequences in
733 sediments sampled during the course of the experiment (T0-T7) in the surface (s) and
734 deep (d) layers for both the 16S rRNA transcripts (RNA) and the 16S rRNA genes (DNA).

735

736 **Fig. 6.** Variation in functional gene sequence numbers expressed as z-scores during the
737 course of the experiment (T2-T6). Sequence counts were obtained after annotating
738 metagenomes against the KEGG orthology database.

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