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HAL Authorization

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## Significant SAR11 removal by a hard-bottom community

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18

19            **Abstract**  
20

21            Microorganisms are a key component of the marine food webs through the microbial  
22 loop. In previous work, we have shown that some bacteria, including *Candidatus*  
23 *Pelagibacter* spp (SAR11)—the most abundant bacterium in the ocean—can evade filtration  
24 by benthic and planktonic tunicates. Here we tested whether differential removal of microbial  
25 taxa by benthic filter-feeders can be observed in the distribution and abundance of microbial  
26 taxa from hard-bottom subtidal communities, a common coastal habitat in the Eastern  
27 Mediterranean Sea towards the open sea. The abundance of microbial groups along cross-  
28 shore transects was measured based on combined flow cytometry and SSU rRNA gene  
29 metabarcoding. Our results show that most groups were depleted (up to 50%) over the hard-  
30 bottom compared to the open sea, but unexpectedly we did not observe a clear differential  
31 removal of different taxa, SAR 11 notably. This study indicates a strong top-down control of  
32 the abundance of pelagic microorganisms over shallow hard-bottom where suspension  
33 feeders are common.

34

## 35 Introduction

36

37 Marine microbial communities form the basis of the ocean food web and mediate  
38 most of the energy and material fluxes in the ocean (Glöckner et al. 2012). Microorganisms  
39 constitute a large fraction of the living biomass in the sea (Pomeroy et al. 2007), and the  
40 structure and function of their populations are shaped by a delicate balance between growth  
41 and mortality (Pernthaler 2005). Grazing and virus-driven lysis constitute the main sources of  
42 mortality (Sánchez et al. 2020), countered by the capabilities of microorganisms to avoid  
43 grazing (Matz and Kjelleberg 2005), survive digestion or resist viral lysis. Grazing or  
44 predation on microorganisms by either planktonic or benthic organisms is an important  
45 mortality factor in many habitats (e.g., Verity 1991; Gili and Coma 1998; Gorsky et al. 1999;  
46 Riisgard and Larsen 2001; Pernthaler 2005; Patten et al. 2011a). In the pelagic realm, protists  
47 commonly dominate the guild of grazers of bacteria (Calbet and Landry 2004; Matz and  
48 Kjelleberg 2005). However, in some habitats and seasons, grazing by metazoan and lysis by  
49 phages may dominate mortality (Hahn and Höfle 2001).

50 Bacteria form some 65-86% of the biomass of microorganisms in the upper ocean  
51 (Morris et al. 2002). However, only a small number of bacterial groups dominate that guild  
52 (Teeling et al. 2012). In the oligotrophic eastern Mediterranean, pico-cyanobacteria  
53 (*Synechococcus* and *Prochlorococcus*) and a few members of *Candidatus Pelagibacter*  
54 *ubique* (SAR11) clade dominate microbial communities, accounting for >70% of the total  
55 bacterial biomass (Partensky et al. 1999, Dadon-Pilosof et al. 2017). SAR11 is a clade of  
56 heterotrophic bacteria, which constitutes 15–60% of total bacteria in the upper ocean (Morris  
57 et al. 2002,2012; Rappé et al. 2002; Eiler et al. 2009; Giovannoni 2017). It is one of the  
58 smallest free-living bacteria in the sea and is thought to be the most abundant group in the  
59 world ocean. Within that group, some ecotypes thrive in oligotrophic environments while  
60 others in more productive waters (Morris et al. 2002, Salter et al 2015). Members of the

61 SAR11 clade also have the lowest nucleic acid content (LNA) among all non-photosynthetic  
62 bacteria (Mary et al. 2006). Dadon-Pilosof et al. (2017) reported that some bacteria,  
63 especially members of the SAR11 clade, can effectively evade grazing from both pelagic and  
64 benthic tunicates and this lack of grazing pressure on SAR11 could partially explain its  
65 abundance and ubiquity.

66 A diverse guild of benthic invertebrate suspension feeders, including sponges,  
67 bivalves, cirripedians cnidarians, bryozoans, and tunicates, often dominates subtidal hard  
68 substrates (Topçu et al. 2010). Their diet ranges from consumption of dissolved organic  
69 matter (DOM) through grazing on microorganisms such as phytoplankton, virioplankton,  
70 archaea and bacteria, as well as feeding on zooplankton and detritus (Gili and Coma 1998;  
71 Topçu et al. 2010). Hard-bottom subtidal communities along the Mediterranean Sea are  
72 diverse and are undergoing dramatic changes in the recent decades due to the combined effect  
73 of global warming, overfishing, and the introduction of invasive species ( Rilov et al. 2019).  
74 Within these communities, retention of small particles is an adaptive advantage since  
75 picoplankton often dominate the planktonic community biomass (Topçu et al 2010).

76 Grazing by benthic suspension feeders on microorganisms is also an important  
77 component of the benthic-pelagic coupling in coral reefs (Yahel et al. 1998; Genin et al.  
78 2002, 2009; Patten et al. 2011). This grazing pressure on the microbial community is not  
79 necessarily uniform as it depends on the spatial heterogeneity of the distribution of different  
80 suspension feeders and their respective diets (Yahel et al. 2006, 2009; Hanson et al. 2009;  
81 Dadon-Pilosof et al. 2017). Differential capture of particles from the ambient water based on  
82 their size, concentration or morphological features is therefore expected to be reflected in the  
83 prey distribution (Gili and Coma 1998).

84 The goal of the current study was to indirectly evaluate the effect of the whole benthic  
85 community removal on the distribution of microorganisms across a shallow, subtropical

86 rocky coast. Following the study of Yahel et al. (1998) across a coral reef, a study that  
87 preceded the demonstration of the role of phytoplankton grazing in the trophic dynamics of  
88 coral reefs (Genin et al. 2009; Monismith et al. 2010), we sought to test the hypothesis that  
89 differential removal on microbes is also reflected in the cross-shore distribution of bacteria  
90 and other picoplankton.

## 91        Methods

### 92        Study site

93        Sampling was conducted off Michmoret, Israel (32° 24'N, 34° 52'E) in the Eastern  
94 Mediterranean Sea. The oligotrophic Israeli shoreline is characterized by extremely low  
95 nutrient levels (Nitrate 0.5-1  $\mu$ M, Phosphate 0.05-0.1  $\mu$ M, Krom and Suari 2015), high  
96 salinity (38.3 to 40.0 PSU) and relatively warm waters (16.5 to 30.8°C) (Suari et al. 2019)  
97 that is dominated in terms of numbers and biomass by pico- and nanoplankton sized  
98 organisms (Herut et al. 2000, Raveh et al. 2015). Water stratification appears usually in  
99 spring following a deep winter mixing that is enhanced by the high salinity of surface waters.

100        Eight transects were performed between December 2015 and April 2017 (Table S1).  
101 Each transect encompassed eight stations spanning from 1.5 m depth at the hard-bottom  
102 subtidal towards the “open-sea” (~ 0.9 km off-shore, water depth >12 m, Fig. 1). During the  
103 sampling periods the average temperature ( $\pm$ SD) was 19.7 $\pm$ 0.7 °C, salinity was high  
104 (39.25 $\pm$ 0.2 PSU), and the water column was fully oxygenated (dissolved oxygen  
105 concentration 216 $\pm$ 6  $\mu$ M). Due to the need to work very close to the bottom at the rocky  
106 shoreline, sampling dates were dictated by sea conditions and were limited to days of calm  
107 sea (wave height < 30 cm) and weak winds (<6m s<sup>-1</sup>). To get a larger scale context within the  
108 eastern Mediterranean shelf offshore of the high-resolution cross-shore transects, six cross-  
109 shelf transects were sampled during March 2018, spanning from one km offshore, equivalent

110 to the farthest station in the small scale transects, to 42 km west to the shelf edge at 750 m  
111 bottom depth (Fig S1).

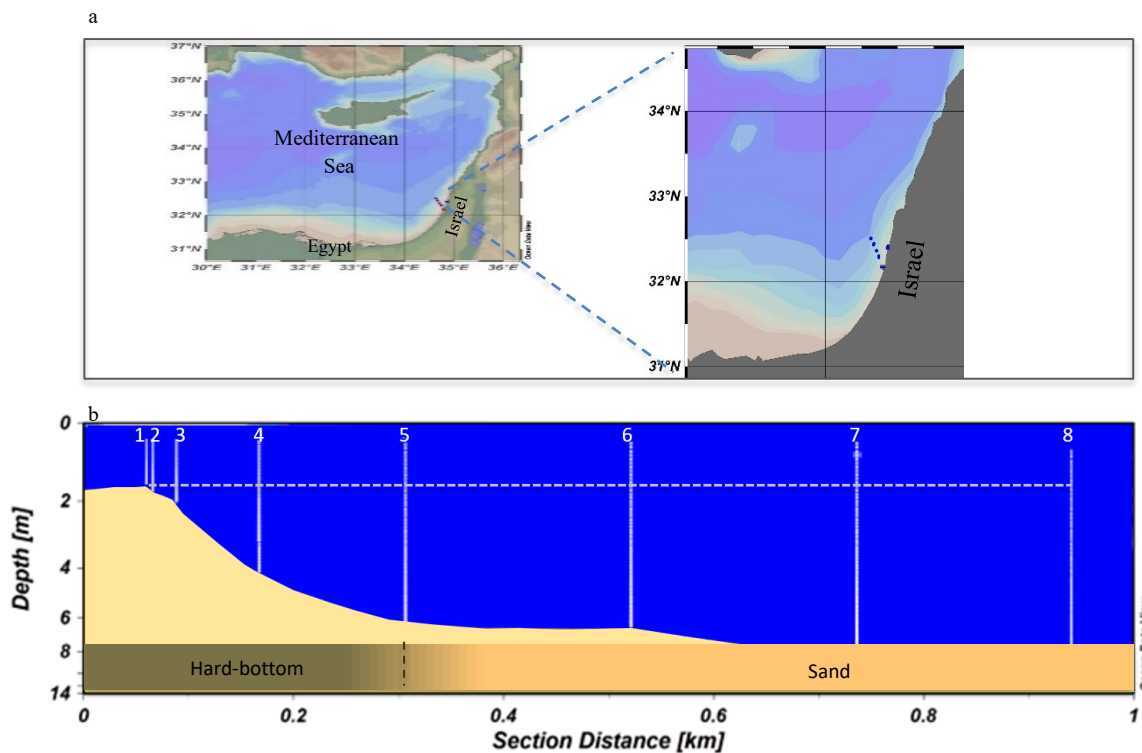


Figure 1: The location of the transect line in the Eastern Mediterranean (a) transects location along the Israeli shore (b) transect showing the bottom (yellow), sampling stations (white numbered lines), dotted white line (1.5 m depth) representing sampling depth.

112  
113 The shallowest sampling station in each transect was located ~150 m offshore at 1.6-  
114 1.8 m depth where Niskin bottle was placed just above the rocky bottom (c.a. 10 cm) to  
115 collect a sample. The 1.5 m sampling depth was retained along the transect as the water  
116 deepened down to 12 m at the furthest offshore station, ~0.9 km offshore. The four  
117 shallowest stations (hereafter Stations 1-4) were located above hard bottom (rocky area), the  
118 5<sup>th</sup> station was located at the boundary between rocks and sand, while stations 6 to 8 were  
119 above sandy bottom.

## 120 Sampling methods

121 Prior to sampling, each station was marked with a moored surface buoy and profiles  
122 of temperature, salinity, fluorescence, and oxygen were measured using a Seabird

123 SBE19plusV2 CTD equipped with an *in vivo* chlorophyll fluorimeter (Cyclop7, Turner  
124 Designs) and a dissolved oxygen sensor (SBE43, Seabird). The CTD was lowered in a  
125 horizontal position at a slow pace (about a meter per minute) so that the water column was  
126 adequately profiled even in the shallow-most stations. Due to the need to work close to the  
127 bottom at the rocky shoreline, sampling was conducted from either a kayak or a small  
128 inflatable skiff.

129         Seawater was collected simultaneously with CTD profiles, using a 5 L Niskin bottle.  
130 At each station, samples were collected directly from the Niskin bottle for all the required  
131 analyses: flow cytometry, DNA extraction and Chl-a measurements. Water samples were  
132 kept on ice in the dark for further processing in the laboratory within 2-3 hours. Samples for  
133 DNA were collected only during three of the eight sampling times.

#### 134         Sample analysis

##### 135             Flow cytometry

136         Flow cytometry was used to quantify the concentrations and the cell characteristics of  
137 non-photosynthetic microorganisms (hereafter referred to as non-photosynthetic bacteria),  
138 and the following four dominant autotrophic groups: *Prochlorococcus* (Pro), *Synechococcus*  
139 (Syn), pico-eukaryotic algae (PicoEuk), and nano-eukaryotic algae (NanoEuk). We used an  
140 Attune® Acoustic Focusing Flow Cytometer (Applied Biosystems) equipped with a syringe-  
141 based fluidic system that allows a precise adjustment of the injected sample volume and  
142 hence high precision of the measurements of cell concentrations ( $\pm 5\%$ ). The instrument's  
143 optical unit contained violet and blue lasers (405 and 488 nm, respectively) and was further  
144 adapted for the analysis of marine ultra-plankton samples as described below.

145         Aliquots of 1.8 mL were collected from each water sample and transferred into 2 mL  
146 cryotubes (Corning cat No. 430659). Samples were first incubated for 15 min at room  
147 temperature with Glutaraldehyde 50% (electron microscopy grade, Sigma-Aldrich, cat No.

148 340855) at 0.1% (final concentration). Samples were frozen in liquid nitrogen (at least 60  
149 min) and then stored at -80 °C until analysis (within a few days).

150 Each sample was analyzed twice. First, 600  $\mu$ l of the sample water was analyzed at a  
151 high flow rate (100  $\mu$ L min<sup>-1</sup>) for the determination of ultra-phytoplankton with a dual  
152 threshold (trigger) on the red fluorescence channels of the violet and blue lasers. A second  
153 run was used to analyze cells with no autofluorescence, i.e., non-photosynthetic microbes. To  
154 visualize these cells, a 300  $\mu$ L aliquot of the sampled water was incubated with the nucleic  
155 acid stain SYBR Green I (20-120 min dark incubation at room temperature, 1:10<sup>4</sup> of the  
156 SYBR Green commercial stock). For this run, we used a low flow rate of 25  $\mu$ L min<sup>-1</sup> and the  
157 instrument was set to high sensitivity mode. Seventy-five  $\mu$ L of the sample water was  
158 analyzed with a dual threshold (trigger) on green fluorescence channels of the violet and blue  
159 lasers. The taxonomic identification was based on orange fluorescence (B12, 574 $\pm$ 13 nm) of  
160 phycoerythrin and red fluorescence (B13, 690 $\pm$ 20 nm and VL3, 685 $\pm$ 20 nm) of chlorophylls;  
161 side-scatter (SSC), provided a proxy of cell surface complexity and cell volume (Marie et al.  
162 1999), and forward-scatter (FSC) was a proxy of cell size (Cunningham and Buonaccorsi  
163 1992; Simon et al. 1994).

164 Where possible, the non-photosynthetic bacteria were further divided based on their  
165 green fluorescence (proxy for nucleic acid content) and forward scatter (proxy for size) into  
166 three groups: LNA, low nucleic acid non-photosynthetic bacteria; HNA-Ls, high nucleic acid  
167 low-scatter non-photosynthetic bacteria; HNA-Hs, high nucleic acid high-scatter non-  
168 photosynthetic bacteria (Zubkov et al. 2004). Similarly, the eukaryotic algae were separated  
169 to pico- and nano-phytoplankton (Simon et al. 1994). The size of *Synechococcus* is still  
170 somewhat controversial, indicating a range of 0.3 to 1.2  $\mu$ m (e.g., Uysal 2001; Garcia et al.  
171 2016). For pico-eukaryotic algae, we followed Worden and Not (2008), who suggested a size  
172 range of up to 3.0  $\mu$ m. Larger cells were termed nano-eukaryotic algae (2.0-20  $\mu$ m). As a

173 rough proxy of cell size, we used the ratio of the median forward scatter of each cell  
174 population to that of the median forward scatter of reference beads (Polysciences™, cat#  
175 23517, Flow Check High-Intensity Green Alignment 1.0 µm) that were used as an internal  
176 standard in each sample. See Dadon-Pilosof et al. (2019) for a further discussion of the  
177 accuracy of size estimates.

#### 178 Chlorophyll measurements

179 Water samples (~300 mL) were collected directly from the Niskin bottles at each  
180 station into dark volumetric BOD glass bottles (Wheaton 227667) and maintained on ice in  
181 dark cool box. In the lab, samples were prefiltered through a 100 µm mesh (to remove large  
182 zooplankton and/or aggregates and suspended pieces of benthic algae) and filtered using low  
183 vacuum onto a 25 mm Whatman GF/F filter. Filters were kept frozen at -20°C in 20 mL  
184 scintillation vials until further processing. To ensure complete chlorophyll *a* extraction of  
185 coastal phytoplankton, we used a hot dimethyl sulfoxide (DMSO) extraction method  
186 (Burnison 1980). Briefly, 2 mL of DMSO were added into each vial containing the frozen  
187 filter. Vials were then incubated for 20 min at 65°C, then cooled in a dark box to room  
188 temperature (approximately 1 hr). Four mL of buffered Acetone (90% Acetone, 10%  
189 saturated MgCO<sub>3</sub>) were added to the vial and thoroughly mixed. Vials were then left to settle  
190 for few minutes and 3 mL sample was drawn from the vial to a fluorometer cuvette.  
191 Fluorescence was measured with a calibrated Trilogy fluorometer (Turner Designs) using  
192 the non-acidification method (Welschmeyer and Naughton 1994).

#### 193 DNA extraction

194 The relative abundance of prominent microbial taxa (phylotypes) in the seawater was  
195 estimated using next-generation sequencing (NGS) of SSU rRNA genes to evaluate any  
196 differential removal by suspension feeders benthic community. Ten mL of seawater collected  
197 from each station and filtered on a 25 mm, 0.2 µm polycarbonate membrane (GE Healthcare

198 Biosciences, cat. No. 110606) under low vacuum and frozen in 1.5 mL micro-tubes at -20°C  
199 until analysis. DNA from each filter was extracted using the DNeasy ‘blood & tissue kit’  
200 (QIAGEN, Cat. No. 69504) with the following modifications to the manufacturer’s protocol:  
201 ATL buffer (180 µL) and 20 µL of proteinase K were added and samples were incubated at  
202 56°C for 1 hr. Then 200 µL of AL Buffer and 200 µL of 95-100% ethanol was added to the  
203 sample and the mixture was pipetted into spin columns and placed in a 2 mL collection tube.  
204 Tubes were centrifuged at 6000 RCF for 1 min. The flow-through was discarded and 500 µL  
205 of AW1 buffer was added to the column, centrifuged at 6000 RCF for 1 min, and the flow-  
206 through again discarded. This step was repeated for the third time, with 500 µL Buffer AW2  
207 and a spin of 18,000 RCF for 1 min to dry the membrane before elution. For the elution step,  
208 the spin column was placed on a new collection tube. Two hundred µL of buffer AE  
209 preheated to 56°C was pipetted at three steps (50 µL, 50 µL, and 100 µL) into the column and  
210 each step was followed by 6000 RCF centrifugation for 1 min. The sample was then  
211 incubated at room temperature for at least a minute and stored at -20°C.

## 212 [Next-generation sequencing](#)

213 Samples were sequenced by Research and Testing Laboratories (Lubbock TX). The  
214 SSU rRNA genes were amplified for sequencing using a forward and reverse fusion primers  
215 (515F-Y - 926R (Parada et al. 2016)). The forward primer was constructed with (5’-3’) the  
216 Illumina i5 adapter (AATGATACGGCGACCACCGAGATCTACAC), an 8-10bp barcode, a  
217 primer pad, and the 5’- GAGTTTGATCNTGGCTCAG -3’ primer. The reverse fusion  
218 primer was constructed with (5’-3’) the Illumina i7 adapter  
219 (CAAGCAGAAGACGGCATAACGAGAT), an 8-10bp barcode, a primer pad, and the 5’-  
220 GTNTTACNGCGGCKGCTG -3’ primer. Primer pads were designed to ensure the primer  
221 pad/primer combination had a melting temperature of 63°C-66°C, according to methods  
222 developed by Patrick Schloss' laboratory ([9](http://www.mothur.org/w/images/0/0c/Wet-</a></p></div><div data-bbox=)

223 lab\_MiSeq\_SOP.pdf). Amplifications were performed in 25  $\mu$ L reactions with Qiagen  
224 HotStar Taq master mix (Qiagen Inc, Valencia, California), 1  $\mu$ L of each 5  $\mu$ M primers, and 1  
225  $\mu$ L of the template. Reactions were performed on ABI Veriti thermocyclers (Applied  
226 Biosystems, Carlsbad, California) under the following cycle conditions: 95°C for 5 min, then  
227 35 cycles of 94°C for 30 sec, 54°C for 40 sec, 72°C for 1 min, followed by one cycle of 72°C  
228 for 10 min and a final 4°C hold.

229 Amplification products were visualized with eGels (Life Technologies, Grand Island,  
230 New York). Products were then pooled equimolarly and each pool was size-selected in two  
231 rounds using Agencourt AMPure XP (BeckmanCoulter, Indianapolis, Indiana) in a 0.7 ratio  
232 for both rounds. Size-selected pools were then quantified using a Qubit 2.0 fluorometer (Life  
233 Technologies) and loaded on an Illumina MiSeq (Illumina, Inc. San Diego, California) 2x300  
234 flow cell at 10 pM.

#### 235 Sequence data analysis

236 SSU rRNA sequences were treated using a qiime2 (v. 2018-8) and biom-format (v.  
237 2.1.6) deployed through a bash scripts (Suppl. File). Briefly demultiplexed forward and  
238 reverse reads were imported into qiime2 artifacts and ASV tables were generated using *qiime*  
239 *dada2* using options "--p-trim-left-f 19 --p-trim-left-r 20 --p-trunc-len-f 300 --p-trunc-len-r  
240 250" The resulting ASVs were identified using *qiime feature-classifier* using an in house  
241 version of the Silva132 (arb-silva.de) including only the region flanked by the primers, and a  
242 taxonomy files extracted from this database (see data availability below for details).

243 Since the primers we used amplify both 16S rRNA from prokaryotes and chloroplasts  
244 as well as 18S rRNA from eukaryotes, the ASV table and sequence files were filtered using  
245 *qiime taxa* to generate three sets of table/sequences, one with 16S rRNA, one with  
246 chloroplasts and one with eukaryotic sequences. ASV tables were modified to include

247 taxonomy using *sed* and *gawk* and *biom-format* and exported in ".tsv" format for further  
248 analyses. Here we describe only results concerning 2388 prokaryotic ASVs.

#### 249 Data analysis

250 Due to temporal changes in microbial communities, and assuming the entire transect  
251 represented a single water mass the concentrations along each transect were normalized to the  
252 seaward-most station and presented as “% of open sea”. The significance level of cross-shore  
253 trends was tested using the “Page test” for ordered alternatives (Page 1963). This non-  
254 parametric test is a modified version of the Kruskal-Wallis one-way ANOVA for ranked data.  
255 Nearshore depletion of microbial taxa was tested (each taxon per season) with  $H_1$  as an  
256 ordered decrease in concentrations from the “open water” toward the shore. Due to missing  
257 sampling points, only 3 complete transects toward the hard-bottom were used for the test in  
258 each season.

259 Data collected with the CTD was converted, aligned, and binned (at 0.1 m) with the  
260 SBE DataProcessing software (Version 7.2). Since salinity differences along the transects  
261 were negligible (<0.01 PSU), density differences were driven solely by temperature. The  
262 vertical and horizontal change in temperature and *in-vivo* chlorophyll fluorescence along the  
263 cross-shore transects (hereafter ‘anomaly’) was calculated within each transect as anomalies  
264 (the difference of each data point from the average temperature or fluorescence along the  
265 transect). All seasonal anomalies were plotted together using Ocean Data View (Version 5).  
266 Interpolation was made with the Weighted-average gridding function of Ocean Data View  
267 using a seeking distance of 0.25 m along the vertical axis and 100 m along the horizontal  
268 axis. Due to the low (N=4), data are presented as a seasonal average  $\pm$  standard error (SE)  
269 unless otherwise indicated.

270 Removal of specific ASVs toward the shore was calculated by multiplying the relative  
271 abundance of each ASV and the total bacterial cell counts obtained by flow cytometry and

272 then calculation as a percentage of the open sea station. Normalized removal was calculated  
273 by normalizing measured removal to the ASV with the highest removal within the same  
274 transect. Implementation of this approach provides a powerful tool to indirectly evaluate the  
275 effect of benthic removal of microbial prey at the ASV level. Hereafter, the use of the terms  
276 “selectivity” and “preference” are limited to their technical definition (Chesson 1978, 1983),  
277 i.e., the removal of a prey type in higher proportion than its proportional presence in the  
278 environment, relative to other food types present.

#### 279 **Data availability**

280 Raw sequencing reads are available through the NCBI SRA under accession number  
281 PRJNA912166. The analysis pipeline and associated files including scripts, mapping files,  
282 taxonomic identification databases and ASV tables are available through a github repository  
283 ([github.com/suzumar/transect\\_ms](https://github.com/suzumar/transect_ms)).

#### 284 **Results**

285 During winter, a temperature gradient was found along the transects with colder water  
286 at the shallow stations above the hard-bottom (stations 1-5,  $19.4 \pm 0.03^\circ\text{C}$ , Fig 2a) in  
287 comparison to the "open sea" (stations 6-8,  $20.1 \pm 0.03^\circ\text{C}$ , Fig 2a). During spring, no such  
288 temperature gradient was observed along the transects but the beginning of stratification was  
289 noticeable, with a half-degree Celsius warmer surface layer (0-3 m,  $20.1^\circ\text{C} \pm 0.07$ , Fig 2b)  
290 than the deeper water (3-12m,  $19.6^\circ\text{C} \pm 0.02$ , Fig 2b). The warmer, nearshore surface layer  
291 (10-25 m depth, Fig S1) showed typical coastal enrichment, with higher chlorophyll  
292 concentrations (Fig S1) and cell counts (Fig S2) than the open sea. Salinity was similar along  
293 the transect (1.5m depth) and with depth (0-12m) during winter and spring (39.26-39.31 PSU  
294 and 39.35-39.28 PSU, respectively, during winter and, 39.29-39.23 PSU and 39.06-39.21  
295 PSU, respectively, during spring).

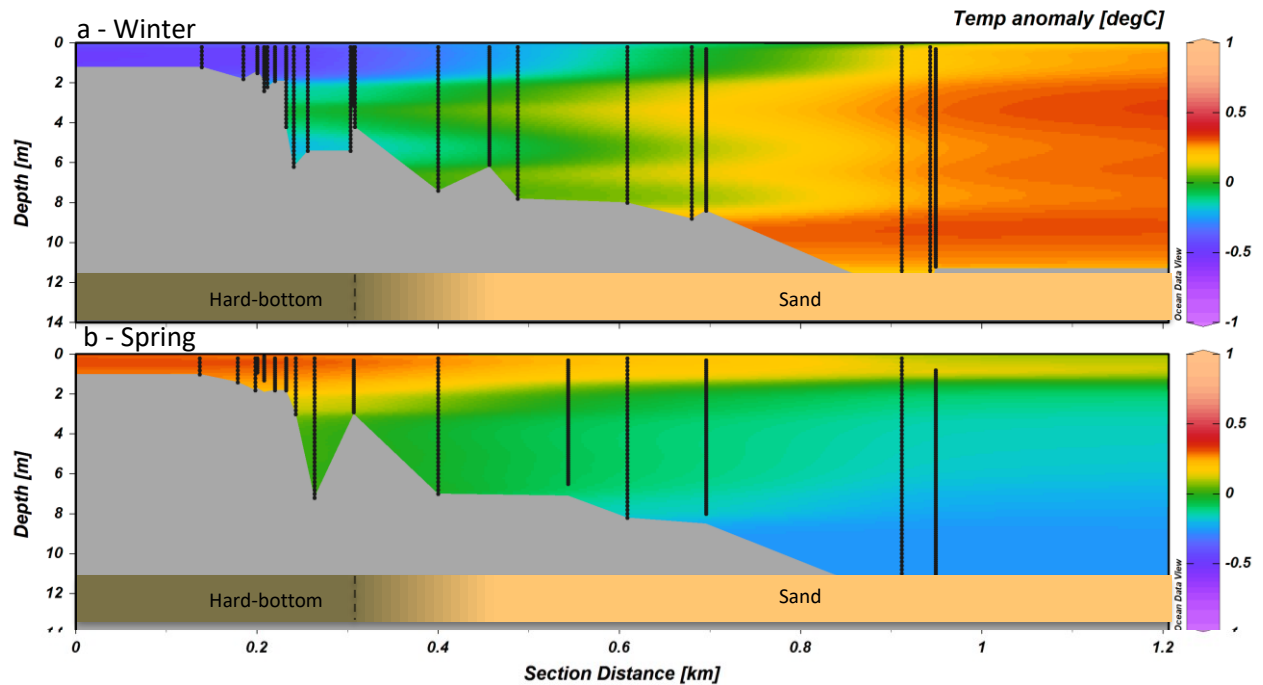


Figure 2: Temperature anomaly along the cross-shore transects. (a) Winters of 2015-2016, n=4. (b) Springs of 2016-2017, n=4. Sampling stations are shown as vertical black lines. Grey shading approximates the bottom depths based on the bottom depth of each cast. In most cases, the CTD was lower all the way to the bottom (~0.2 mab).

296 In the cross-shore transects, the concentration of all microbial populations decreased  
297 toward shore in a highly significant gradient (Page test,  $p < 0.01$ , Fig. 3a-d). In most cases, and  
298 for most of the microbial populations, cell concentrations were similar along the ~700 m  
299 sandy section of the transects, between the open sea and the outer boundary of the rocky area  
300 (Stations 6 to 8). Sometimes even an increase was observed along that section (Table S1, S2).  
301 Most of the depletion occurred above the rocky habitat (Stations 1 to 5). The depletion of the  
302 microbial populations over the rocky section range between 25-50% of their percentage of  
303 the open sea (Fig 3a-d). *In vivo* chlorophyll fluorescence (Fig S3) and the concentration of  
304 extracted chlorophyll a also showed a strong depletion above the hard-bottom area compared  
305 to the open sea (Fig 3e).

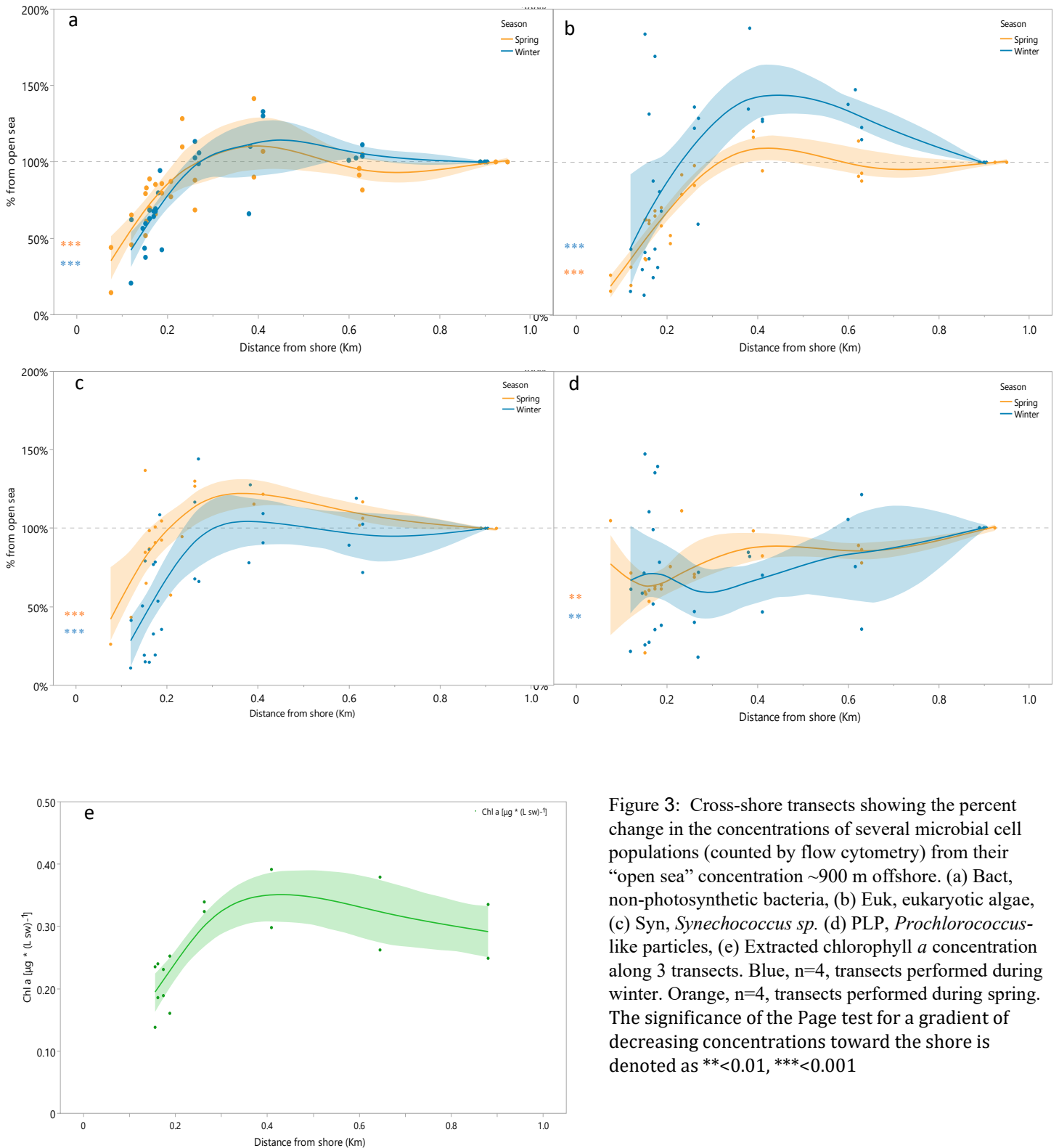


Figure 3: Cross-shore transects showing the percent change in the concentrations of several microbial cell populations (counted by flow cytometry) from their “open sea” concentration ~900 m offshore. (a) Bact, non-photosynthetic bacteria, (b) Euk, eukaryotic algae, (c) Syn, *Synechococcus sp.* (d) PLP, *Prochlorococcus*-like particles, (e) Extracted chlorophyll *a* concentration along 3 transects. Blue, n=4, transects performed during winter. Orange, n=4, transects performed during spring. The significance of the Page test for a gradient of decreasing concentrations toward the shore is denoted as \*\*<0.01, \*\*\*<0.001

341

342 Differential changes in relative abundances between ASVs and even within the same

343 ASV in different transects were observed (Fig 4a-c). For example, ASVs belonging to

344 cyanobacteria group were on average 8%, 35% and 3% of their percentage at the open sea  
 345 (Fig 4a-c accordingly).

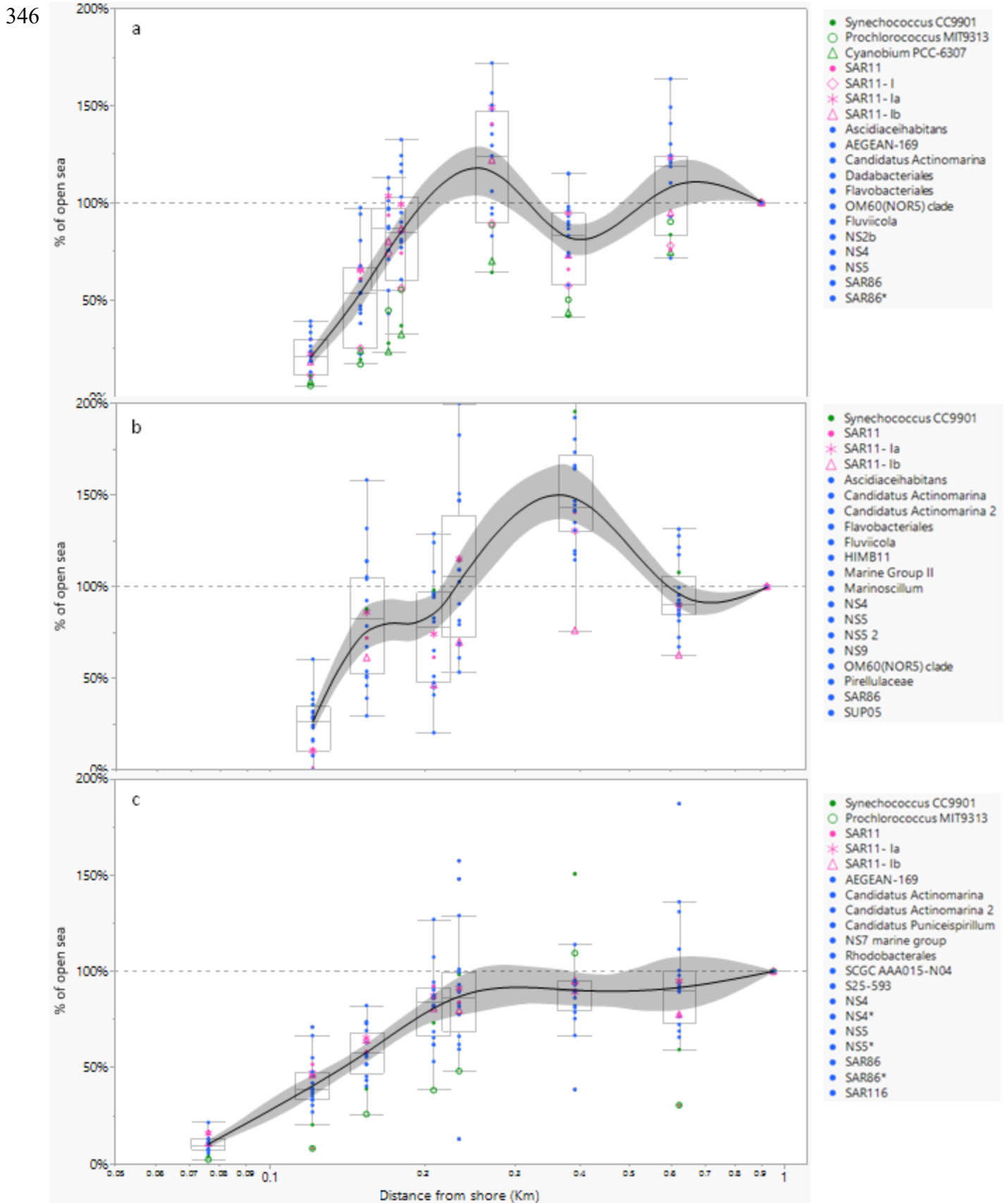


Figure 4: Cross-shore transects showing the percent change in the concentrations of the 20 most abundant prokaryotic ASVs in the water relative to their “open sea” concentration ~900 m offshore: (a-c) transects above rocky hard bottom (January 2016, March 2016 and, April 2016). The vertical dashed line represents the concentration in the open sea (100%). Pink indicates members of SAR11 clade, green indicates autotrophs, and blue indicates other non-photosynthetic bacteria.

347            Within the SAR11 clade, the percentage of four ASVs decreased along the transects,  
348    in similar trends (Fig 5). Transects showed  $83\% \pm 7\%$  (Mean  $\pm$ SD, Fig 6a-c) normalized  
349    removal for the 20 most abundant ASVs. Abundant ASVs including *Synechococcus* CC9901  
350    (27% relative abundance, transect during January 2016) and SAR11-Ia (25%, transect during  
351    April 2016) were removed at similar percentages (95% and 84% accordingly) compared to  
352    less abundant ASVs (e.g. SAR11-Ib 1.3% relative abundance and 87% removal, SAR86  
353    2.3% relative abundance and 86% removal) (Fig 6).  
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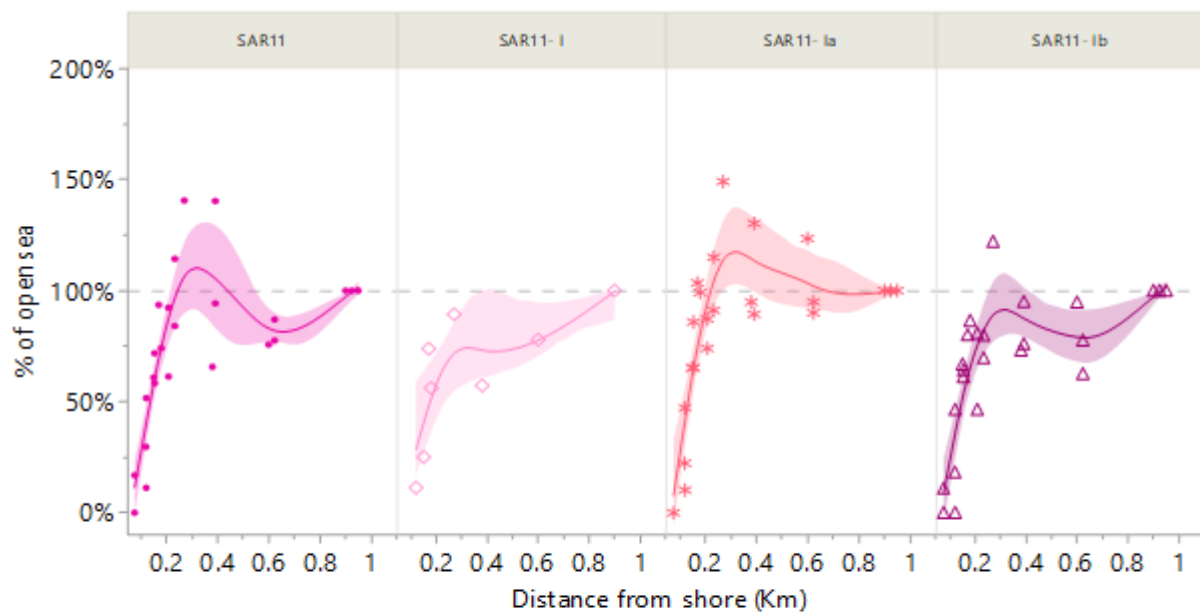


Figure 5: Cross-shore transects showing the percent change in the concentrations of four ASVs belonging to SAR11 clade in the water relative to their “open sea” concentration ~900 m offshore: (a-c) transects above rocky hard bottom (January 2016, March 2016 and, April 2016). The vertical dashed line represents the concentration in the open sea (100%).

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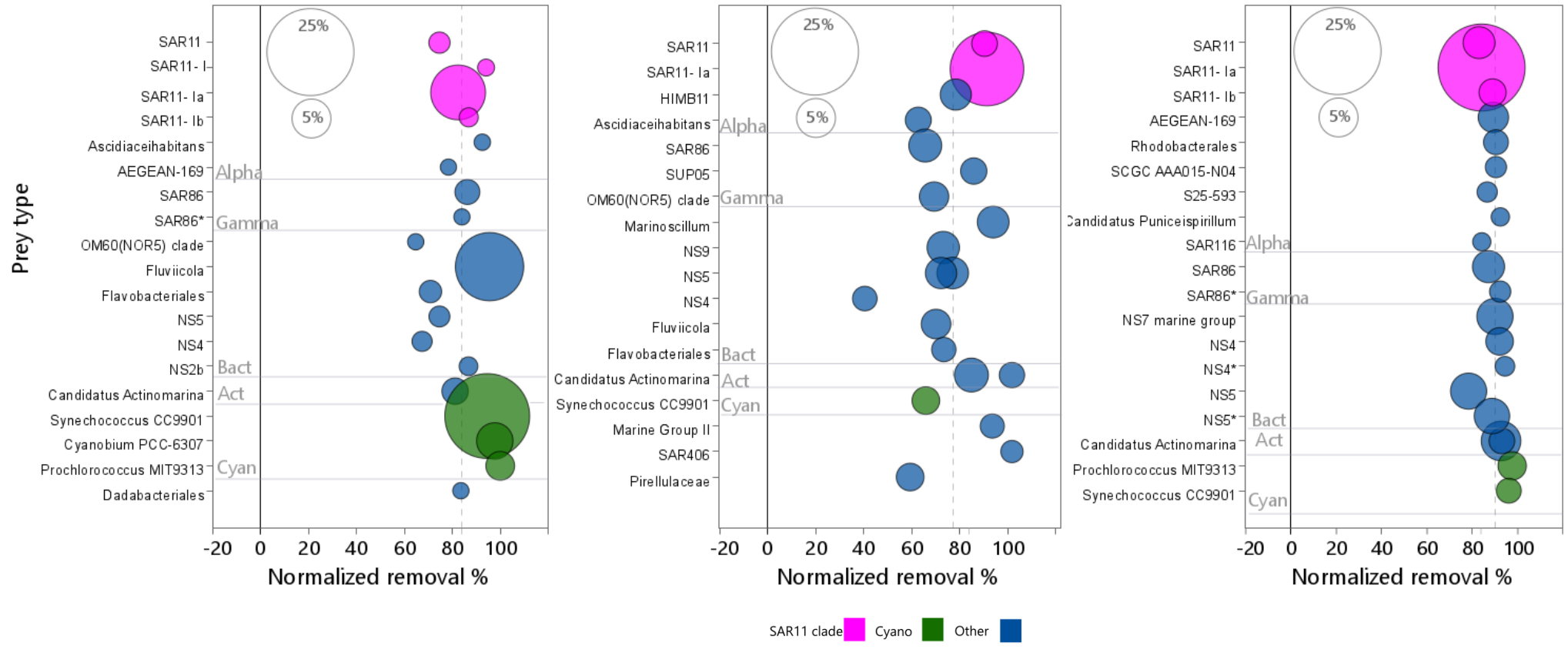


Figure 6: Normalized removal of the 20 most abundant ASVs from the open sea toward the shore. (a-c) transects above rocky hard bottom (January 2016, March 2016 and, April 2016). Pink indicates members of SAR11 clade, green indicates autotrophs, and blue indicates other non-photosynthetic bacteria. Dashed vertical line represents the expected retention assuming equal retention probability for all cells. Size of circles represents relative abundance in the open water (Station 8) during sampling., with the two white circles plotted for scaling.

## Discussion

368  
369 This study was designed to evaluate whether the effect of differential removal of  
370 microorganisms by benthic suspension feeders can be detected at the community level. Most  
371 of the microbial populations were depleted above the hard-bottom area compared to the open  
372 sea. Differential removal was observed at low extent (between and within species). A similar  
373 trend was also observed for chlorophyll *a* concentration, suggesting the formation of a  
374 depleted boundary layer over the hard-bottom. The hard-bottom community at the study site  
375 included a diverse assemblage of suspension feeders, including sponges, bivalves, ascidians,  
376 polychaetes, hydrozoans, and bryozoans (Rilov et al. 2018). Suspensions feeders on the hard-  
377 bottom occur in different densities, and hence the competition for available prey is between  
378 species, within species and even between different taxa. Niche speciation is expected in such  
379 diverse and dense community where different taxa utilize different filtration mechanisms and  
380 presumably, different organic carbon sources in their diet. If the depletion of microbial cells  
381 is the outcome of grazing by benthic suspension feeders, near-bottom depletion should  
382 generate a shore-wise depletion above the hard-bottom section of the transects compared to  
383 the sandy section of the transect (e.g., Genin et al. 2009; Jones et al. 2009). In case other  
384 processes determine the cross-shore trend (e.g., runoff, eutrophication, and coastal pollution),  
385 the depleted zone is expected to extend throughout the water column and no difference is  
386 expected along the transects. Differential grazing of phytoplankton by dense populations of  
387 benthic suspension feeders was also reported in San-Francisco bay where it was attributed to  
388 different sinking rates of the microalgae (Lucas et al. 2016). Such a phenomenon is very  
389 unlikely in the east Mediterranean where the planktonic community is dominated by very  
390 small cells (<10  $\mu\text{m}$ ) with negligible sinking rates (Siokou-Frangou et al. 2010). This body of  
391 prior work supports the assumption that the depletion reported here is due to benthic  
392 suspension grazing although it was not measured directly.

393 Physical and biological processes are the major factors controlling changes in particle  
394 concentrations throughout the ocean, and advective processes could be responsible for the  
395 pico and nanoplankton depletion we observed. Monismith et al. (2006) showed that shallow  
396 regions nearshore experience larger temperature changes than deeper regions offshore. When  
397 the water warms during the daytime (e.g., in the spring), the shallow near-shore water body  
398 tends to warm faster than the nearby open sea. The warmer water expands and flows  
399 offshore, causing deeper and cooler water to flow onshore at depth to replace it. This ‘thermal  
400 flow’ (Fig 7) leads to an upwelling of deeper water and material to the nearshore region (Fig  
401 7). The opposite process occurs during winter or cold nights, when a faster cooling of the  
402 shallow, near-shore waterbody initiates near-shore downwelling of cold surface water that  
403 reverses the direction of the “thermal flow” cycle and induces onshore flow of surface water  
404 (Fig 7). In the Eastern Mediterranean Sea, the water column is usually stably stratified, and  
405 the numbers and biomass of surface water plankton is lower than subsurface layers (Suari et  
406 al 2019). Onshore transport of surface water from the offshore during the cold phase of the  
407 thermal flow cycle and the nearshore downwelling of "plankton poor" seawater is expected to  
408 yield a benthic zone of depleted of plankton close to shore (Labiosa et al. 2003). However,  
409 this physical mechanism would not be expected to change the relative abundance of different  
410 microbial groups along the transects, nor should it differentially affect rocky and sandy  
411 sections of the transects. Moreover, during the warm phase of the thermal flow cycle (in  
412 spring for example), local upwelling is expected to bring plankton-rich water onshore,  
413 countering removal, and the benthic zone should be as rich if not richer nearshore. Our results  
414 showed that nearshore depletion occurred during both the cold and hot ‘phase’ of the thermal  
415 flow, suggesting that a biological process such as filtration is probably the mechanism  
416 responsible for the depletion above the hard-bottom section in the transects rather than  
417 advection of planktonic poor water. A similar conclusion was reached by Patten et al. (2011),

418 who showed depleted levels (~40% on average) of microbial cells over a reef with negligible  
419 removal over a sandy bottom.

420 Our previous work showed partial grazing resistance of members SAR11 clade to  
421 grazing by pelagic and benthic tunicates (Dadon-Pilosof et al. 2017), and in general, lower  
422 retention efficiencies on LNA bacteria than other prey types. Selective grazing and  
423 specifically low retention on LNA compare to both HNA bacteria and *Synechococcus* was  
424 also observed in sponges (Hanson et al. 2009; Jiménez 2011) and preferential retention of  
425 *Synechococcus* and eukaryotic algae over other prey types was observed in bivalves (Yahel et  
426 al. 2009). The results (although using indirect measurements) of this study suggested  
427 inversely to what we expected that members of SAR11 were grazed at higher efficiencies  
428 than other available prey (Fig 5,6) somewhat reflecting their overall high abundance. This  
429 work strengthens the niche speciation assumption, hinting that some taxa within the hard-  
430 bottom community specialize on grazing SAR11. Besides the obvious members of the  
431 suspension feeding community, we must consider the heterotrophic nanoflagellates attached  
432 to the rocks (Yahel et al. 2006). This cryptic community has a large grazing effect that may  
433 be part of the explanation for prey depletion above the hard-bottom. It is known that  
434 heterotrophic nanoflagellates are important bacterivores in pelagic waters (Tophøj et al.  
435 2018).

436 The apparent lack of grazing evasion by members of SAR11 could be possibly  
437 explained by a different mode of interaction to other filter feeders compared to tunicates.  
438 Suspension feeders have different filtration organs, using cilia, mucus, or both to capture and  
439 process suspended particles. While some are active suspension feeders and specialize in  
440 filtration of small particles, others are passive feeders and specialize in filtration of large  
441 particles (Gili and Coma 1998). Different grazing strategies of benthic suspension feeders  
442 would potentially increase the opportunities for exploitation of available prey by

443 communities of suspension feeders (Gili and Coma 1998). Specialization of different  
444 suspension feeders on different prey would explain the homogeneous decrease of different  
445 available prey. While removal by benthic filter feeders is a likely biological explanation for  
446 the depletion of pico- and nanoplankton we observed, other processes such adsorption to  
447 mucus might also have contributed (Decho 1990) although members of SAR11 clade are  
448 known as free-living and not particle-associated or biofilm forming bacteria (Giovannoni  
449 2017, Haro-Moreno et al. 2020)

450 Evaluation of evasiveness using “in versus out” experiments such those in Dadon-  
451 Pilosof et al. (2017) with other taxa, or the measurement of adsorption of different bacteria  
452 including cultured *Candidatus Pelagibacter ubique* to transparent exopolymer particles (Long  
453 and Azam 1996) with water from different points in the transect remain as possible future  
454 experiments to further clarify our results.

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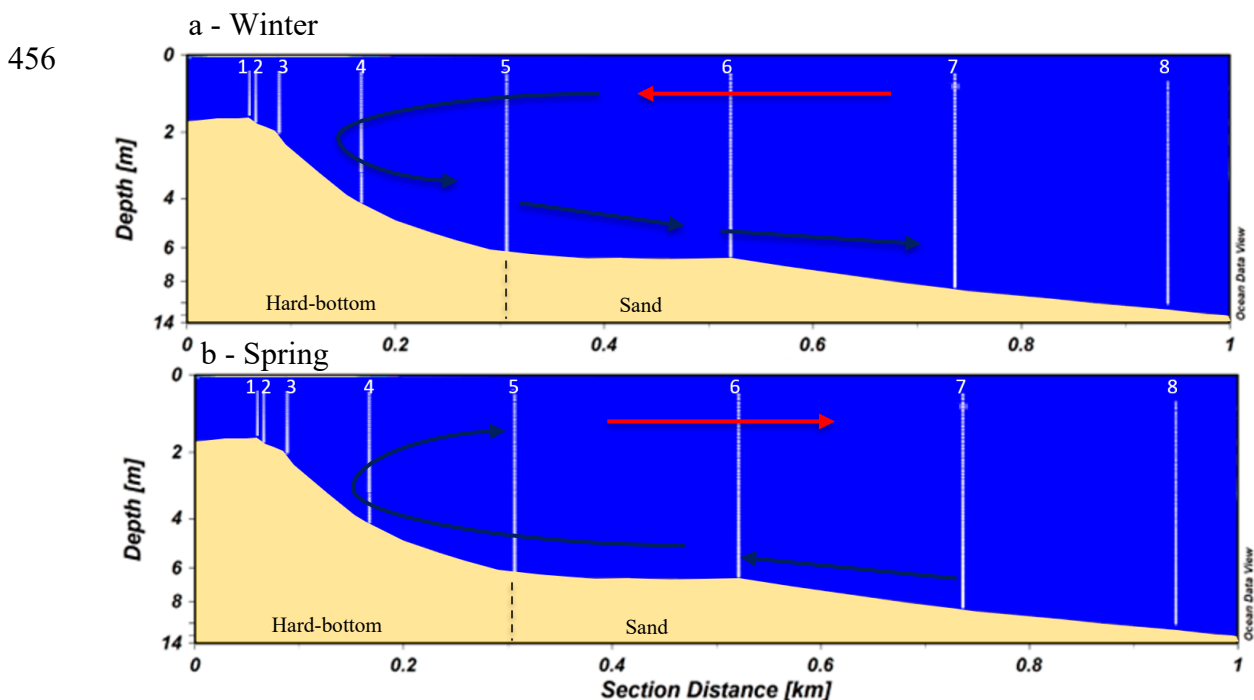


Figure 7: Schematic illustration of a near-shore thermal flow cycle. (a) during winter, when cooling of nearshore water drives offshore flow at depth and onshore flow at the surface. (b) during spring, when the shallow water near shore warms and water flow offshore at the surface and onshore below.

457           Measurements of removal rates of microbial plankton by benthic organisms at the  
458 level of the whole community remain challenging despite of decades of studies of this theme  
459 (Sargent and Austin 1949; Odum and Odum 1955; Johannes et al. 1972). More recent studies,  
460 assessed spatial gradients of DOC, bacterioplankton and virioplankton concentrations in reef  
461 ecosystem resulting in depleted microbial community over the reef compare to negligible  
462 removal over sandy bottom nearby (Patten et al. 2011; Nelson et al. 2011). An alternative  
463 approach utilizes a control-volume approach either by physically enclosing the community in  
464 a bell jar for in situ measurements (e.g., (Hopkinson et al. 1991) or by enclosing and  
465 imaginary “box” over the bottom using vertical arrays of samplers and ADCPs to quantify the  
466 fluxes of plankton into and out of the control volume (Genin et al. 2002, 2009). A third  
467 approach integrates individual rate measurements of dominant benthic suspension feeders  
468 with their abundance and size distribution to assess the community flux and its effect on the  
469 planktonic community in the overlying water (e.g., Genin et al. 2009; Lucas et al. 2016 and  
470 references therein).

471           Further investigation is required to develop and estimate, based on indirect evaluation,  
472 cells removal above hard-bottom suspension feeders. Methodology fine tuning is required for  
473 estimating the cells removal above the hard-bottom compared to open sea and to discriminate  
474 grazing, other biological processes and physical mechanisms. Previous studies showed that  
475 SAR11 evade predation of benthic and pelagic ascidians mucus, but this study showed (albeit  
476 indirectly) that SAR11 was removed efficiently as other prey cells by hard bottom grazers.  
477 Potential grazers might be sponges, bivalves, nanoflagellates attached to the surface of the  
478 rocks which filter their prey by using several different mechanisms. Understanding the  
479 mechanisms and the variance between the grazers' preferences and/or the microbial cells  
480 abilities to evade predation has ramifications on processes affecting the marine food web  
481 such as top-down control and, benthic pelagic coupling. Grazing resistance mechanisms are

482 still understudied and should be investigated further to gain knowledge on its effects in the  
483 marine ecosystem.

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## 677 [Author Contributions:](#)

678 **A.D.P.** designed the study and participated in field experiments, data analysis, and  
679 manuscript preparation. **K.R.C**, participated in data analysis, and contributed substantially to  
680 drafting the manuscript. **M. T.S** designed and performed bioinformatics analyses and  
681 contributed substantially to drafting the manuscript. All authors discussed the results and  
682 commented on the manuscript during its preparation and approved the submitted version of  
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