Microbial community composition and diversity in Caspian Sea sediments

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One sentence summary: This study describes microbial biomass, community composition and diversity in Caspian Sea sediments using lipid and genomic techniques.

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ABSTRACT

The Caspian Sea is heavily polluted due to industrial and agricultural effluents as well as extraction of oil and gas reserves. Microbial communities can influence the fate of contaminants and nutrients. However, insight into the microbial ecology of the Caspian Sea significantly lags behind other marine systems. Here we describe microbial biomass, diversity and composition in sediments collected from three sampling stations in the Caspian Sea. Illumina sequencing of 16S rRNA genes revealed the presence of a number of known bacterial and archaeal heterotrophs suggesting that organic carbon is a primary factor shaping microbial communities. Surface sediments collected from bottom waters with low oxygen levels were dominated by \textit{Gammaproteobacteria} while surface sediments collected from bottom waters under hypoxic conditions were dominated by \textit{Deltaproteobacteria}, specifically sulfate-reducing bacteria. \textit{Thaumarchaeota} was dominant across all surface sediments indicating that nitrogen cycling in this system is strongly influenced by ammonia-oxidizing archaea. This study provides a baseline assessment that may serve as a point of reference as this system changes or as the efficacy of new remediation efforts are implemented.

Keywords: Caspian Sea; marine sediments; bacteria; archaea; Illumina; PLFA

INTRODUCTION

With a volume of 78 000 km\textsuperscript{3} and surface area of $3.8 \times 10^5$ km\textsuperscript{2}, the Caspian Sea is one of the largest inland bodies of water on earth (Dumont 1998). Like the Aral and Black Seas, it was once connected to oceans but has been landlocked for the past five million years. Water depths vary across the Caspian Sea with the northern portion exhibiting a maximum depth of 20 m while the southern portion has a maximum depth of 1025 m (Kosarev and Yablonskaya 1994). Approximately, 130 rivers drain into the Caspian Sea with the Volga River accounting for the majority of...
influents waters (Kosarev 2005). Due to the influx of freshwater, the salinity of the Caspian Sea is approximately one-third of the salinity of seawater making it a lacustrine brackish body of water (Leroy et al., 2007).

Environmental pollution has become a growing problem in this landlocked basin in which contaminants can accumulate and persist. Nutrient-rich industrial and agricultural effluents from rivers have steadily increased over recent years leading to eutrophication (Zomm 2005). Consequently, bottom waters are oxygen deficient and hypoxic zones exist, particularly in the summer months (Diaz 2001). Hypoxic zones can have detrimental effects including mass mortality of benthic organisms, losses in biodiversity and changes in biogeochemical cycles (Diaz and Rosenberg 1995; Wu 2002). The Caspian Sea has also seen substantial expansion of oil and gas exploration and production with the finding of significant reserves, comparable to that of the North Sea (Effimoff 2000). Hydrocarbon concentrations in sediments exceed the background for other fishery water bodies by almost 2-fold (Tolosa et al., 2004). Similarly, concentrations of organochlorinated compounds and heavy metals have been found to exceed sediment quality guidelines (de Mora et al., 2004a, b).

Microbial communities play a pivotal role in the fate of contaminants and nutrients. However, sufficient baseline data is required to establish a foundation to gauge community shifts in response to environmental perturbation or anthropogenic pollution. Sediment microbial communities are particularly important since they have greater cell density and taxonomic diversity than planktonic communities and can respond rapidly to their surrounding environmental conditions (Jørgensen and Boetius 2007; Lozupone and Knight 2007; Gibson et al., 2014). The growth and distribution of sediment microbial communities is strongly affected by the availability of carbon sources and electron acceptors. Persistent hypoxia can lead to changes in the structure and function of microbial communities within sediments (Meyer-Reil and Kostka 2000; Kristiansen, Kristensen and Jensen 2002; Jäntti and Hietanen 2012; Hou et al., 2014).

While there have been studies into the geophysical and hydrological aspects of the Caspian Sea (Peeters et al., 2000; Diaconescu, Kieckhefer and Knapp 2001), the microbial ecology of the Caspian Sea is less well characterized than other marine systems. Previous microbial studies have focused solely on isolating and characterizing oil-degrading microorganisms (Hassan-shahian et al., 2010, Hassanshahian, Emtiazì and Cappello 2012). The primary aim of this study was to describe the biomass, diversity and composition of sediment microbial communities in the Caspian Sea using a combination of lipid and genomic techniques. The secondary aim was to characterize seafloor microbial communities across varying hypoxic conditions to determine how oxygen levels of bottom waters affect microbial community structure. This study is one of the first to survey microbial communities in the Caspian Sea and provides a baseline description that may serve as a point of reference as this system changes. This is particularly important if efforts are undertaken to reduce hypoxia and pollution since shifts in microbial populations are rapid and sensitive indicators of change.

MATERIALS AND METHODS
Site characterization and sample collection

Three sediment cores were collected at three different sampling stations during a research cruise in July 2013 using a Multicorer (Fig. S1, Supporting Information). All sampling stations were located in the southern basin of the Caspian Sea, which has been categorized as persistently hypoxic (<2 mg L⁻¹ of dissolved oxygen) (Diaz and Rosenberg 1995). Temperature, salinity, pH and oxygen concentrations of bottom waters were measured approximately 15 m from the seafloor with a MIDAS CTD + sensor array (Valeport Ltd, St. Peter’s Quay, UK). Stations 1, 2 and 3 had water depths of 600, 205 and 141 m, respectively. Sediment cores ranged in length from 8 to 36 centimeters below seafloor (cmbsf). Following collection, intact cores were sectioned at 4 cm depth intervals and homogenized. Sediment samples were stored at −20 °C on ship and transported to the University of Tennessee for further analysis.

Sediment analysis

Sediment samples were sent to SOEST Laboratory for Analytical Biogeochemistry (University of Hawaii) for total organic carbon (TOC) analyses. Sediment samples were decarbonated with HCl and percentages of total organic carbon (%TOC) were determined using the Shimadzu TOC-L combustion analyzer (Kyoto, Japan). Sediment grain size distributions for each section were determined using a Beckman Coulter LS 230 laser particle size analyzer (Brea, CA, USA) and textures were assessed using the standard textural triangle.

PLFA extraction and analysis

For each sediment sample, 30 to 50 g was extracted using a modified Bligh and Dyer method as per Mahmoudi et al., (2013a). Sediments were extracted using 2:1 methanol/dichloromethane (DCM) and filtered into separatory funnels using 0.45 μm precombusted glass fiber filters (GF/G, Whatman). Following phase separation using nanopure water, the organic phase was collected and separated into three fractions by gravity column chromatography using fully activated silica (precombusted at 450°C for 8 h) and DCM, acetone and methanol to elute non-polar, neutral and polar fractions, respectively. The polar fraction, which contained phospholipids, was evaporated to dryness under a stream of nitrogen gas and reacted to fatty acid methyl esters (FAMEs) via a mild alkaline methanolysis reaction. Identification and quantification of FAMEs utilized an Agilent 6890 gas chromatograph coupled to an Agilent 5973 mass spectrometer (equipped with a 60 m × 0.25 mm Rx×-1 column). The temperature program for the GC oven was 60°C for 2 min, ramp to 150°C at 10°C min⁻¹ and then ramp to 312°C at 4°C min⁻¹. FAMEs were identified using a bacterial reference standard (Bacterial Acid Methyl Esters CP, Mix, Matreya Inc.), mass-fragmentation patterns and retention times and quantified using external calibration standards (which contained FAMEs of various chain length).

Genomic DNA extraction and PCR amplification

Genomic DNA was extracted in triplicate from each sediment sample using the PowerSoil DNA Isolation kit (MoBio Laboratories Inc., Carlsbad, CA, USA) according to manufacturer’s protocol. Triplicate DNA extracts were subsequently further purified using the Genomic DNA Clean and Concentrator kit (Zymo Research, Irvine, CA, USA). 16S rRNA genes were amplified in triplicate using primer pair 515F and 806R (Caporaso et al., 2012). The reverse primers included a 12-bp barcode for multiplexing of samples during sequencing analysis.
Barcoded amplicon sequencing of bacterial and archaeal SSU genes and sequence analysis

16S libraries were prepared according to Caporaso et al., (2012). Briefly, 16S amplicons were pooled together and analyzed by Bioanalyzer (Agilent Technologies) to assess quality and size of amplicons. Following dilution, libraries were subjected to quantitative-PCR to ensure accurate quantification of purified amplicons. 16S libraries were sequenced using an Illumina MiSeq (San Diego, CA, USA) platform at the University of Tennessee. The forward read data (trimmed to 250 bp) was used for analysis due to poor quality and short read length of the reverse reads. Sequences were processed and quality controlled through a combination of the UPARSE and QIIME pipelines (Caporaso et al., 2010a; Edgar 2013). The QIIME (v1.7; Caporaso et al., 2010a) script, split_libraries_fastq.py, was used to demultiplex the sequence data with the quality filter set to zero. The generation of OTUs (97% sequence similarity) and quality control processing was carried out via the UPARSE pipeline (Edger 2013), including de novo and reference-based chimera detection. The resulting OTU table was converted to BIOM format (McDonald et al., 2012). Taxonomy was assigned using the RDP classifier (Wang et al., 2007) against the updated May 2013 ‘13_5/13_8’ Greengenes database (DeSantis et al., 2006; McDonald et al., 2012; Wernery et al., 2012) via QIIME (Caporaso et al., 2010a). A phylogeny was constructed using FastTree (Price, Dehal and Arkin 2010) from a masked PyNAST (Caporaso et al., 2010b) alignment. The resulting phylogeny was manually root to Archaea via Dendroscope (v3; Huson and Scornavacca 2012). Any OTU that comprised less than 0.005% of the total data set was removed to limit the effect of spurious OTUs on analysis (Bokulich et al., 2013; Navas-Molina et al., 2013). Finally, various analyses, evenness and alpha-diversity metrics were calculated after pooling the technical replicates and rarefying the samples to the same sequencing depth using QIIME (Caporaso et al., 2010a), R v3.1 (R Core Team 2013) and Phyloseq (McMurdie and Holmes 2013). After merging replicates, bacterial data were rarefied to ~22 900 reads and archaeal data were rarefied to ~230 reads for non-metric multidimensional scaling (NMDS), principal coordinates analysis and alpha-diversity analysis. For ADONIS analysis individual samples (i.e. all replicates) were rarefied to ~2775 and ~1000 reads for bacteria and archaea, respectively. The sequence data generated in this study were deposited in GenBank under BioProject PRJNA261725 (BioSample Accessions: SAMN03074722-SAMN03074762).

Statistical analyses

Microbial communities across sediment samples were compared using weighted UniFrac (Lozupone and Knight 2005) based on the phylogenetic relationship of representative reads from different sediment samples. Variation in microbial communities over sampling locations and sediment depth was assessed using both a UPGMA tree and ADONIS analysis of all triplicates. Patterns in microbial community structure in relationship to environmental variables were visualized using NMDS plot based on a weighted UniFrac distance matrix. A stress function was used to assess the goodness-of-fit of the ordination. Stress values range from 0 to 1; values below 0.2 suggest that the ordination accurately represents the observed dissimilarity between samples (Clarke 1993). Environmental variables were fitted to the NMDS ordinations as vectors with the ‘envfit’ function of the ‘vegan’ package in R v3.1 (Oksanen et al., 2013).

RESULTS

Environmental parameters of sampling stations and sediment properties

Temperature, pH and salinity of bottom waters (as defined by measurements taken at the bottom meter of a CTD cast) at the three sampling stations were similar; however, considerable variations in oxygen concentrations between stations were observed (Table 1; Fig. S2, Supporting Information). Bottom waters at Station 1 had the lowest concentrations of dissolved oxygen (0.5 mg L\(^{-1}\)), indicative of severe hypoxia. The upper limit for hypoxia in marine environments is approximately 3 mg L\(^{-1}\) of dissolved oxygen (Hofmann et al., 2011). Therefore, bottom waters at Station 2 were mildly hypoxic (3.3 mg L\(^{-1}\)), while bottom waters at Station 3 were under low oxygen saturation (4.8 mg L\(^{-1}\)). Salinity at all three stations was approximately one-third of the mean ocean salinity value of 35 practical salinity units (PSU) (Wilson 1975). TOC varied between sediment cores and was found to be lowest at Station 3 and highest at Station 2 (Table S1, Supporting Information). The TOC values observed at Station 3 were similar to those previously measured in the Caspian Sea (Parr et al., 2007). In contrast, TOC values measured at Station 1 and 2 were higher and consistent with those found in organic-rich, anoxic marine sediments such as those from the Black Sea (Xu et al., 2001). Particle size analysis revealed that the majority of sediment samples were primarily composed of silt and clay particles such that they are classified as having a texture consistent with a silt loam (Table S1, Supporting Information).

Microbial PLFA concentrations and distributions

Microbial biomass, estimated from PLFA concentrations, was found to be highest at Station 2 and lowest at Station 3 (Fig. 1). Using a conversion factor of 5.9 × 10\(^{6}\) cells pmol\(^{-1}\) of PLFA (Mills et al., 2006), PLFA concentrations correspond to cell densities of 1.3 × 10\(^{9}\) to 7.6 × 10\(^{8}\) cells g\(^{-1}\) (Table S2, Supporting Information). Microbial biomass was correlated with TOC; however, some fluctuations were observed at certain depths (Fig. 1). The distribution of PLFAs for all sediments samples was dominated by monounsaturated and n-saturated PLFAs, as expected for sediments (Table S3, Supporting Information) (Zelles 1999). Overall, 16:0 was the most dominant PLFA while 18:0, 11:0 and

<table>
<thead>
<tr>
<th>Station</th>
<th>Latitude</th>
<th>Longitude</th>
<th>Water depth (m)</th>
<th>Temp (°C)</th>
<th>Salinity (PSU)</th>
<th>pH</th>
<th>Oxygen concentration (mg L(^{-1}))</th>
</tr>
</thead>
<tbody>
<tr>
<td>Station 1</td>
<td>39.745</td>
<td>50.480</td>
<td>600</td>
<td>6.1</td>
<td>11.4</td>
<td>7.8</td>
<td>0.5</td>
</tr>
<tr>
<td>Station 2</td>
<td>39.990</td>
<td>51.500</td>
<td>205</td>
<td>6.6</td>
<td>11.3</td>
<td>8.0</td>
<td>3.3</td>
</tr>
<tr>
<td>Station 3</td>
<td>40.040</td>
<td>51.347</td>
<td>141</td>
<td>6.9</td>
<td>11.3</td>
<td>8.1</td>
<td>4.8</td>
</tr>
</tbody>
</table>

Table 1. Coordinates of sampling stations and environmental parameters of bottom waters.
a15:0 were also present. Monounsaturated PLFAs are associated with Gram-negative bacteria (Zelles 1999) such as Proteobacteria, Chloroflexi and Planctomycetes, which were abundant in the sequencing results. Terminally branched PLFA are indicative of Gram-positive bacteria and were present at low levels in all sediment samples (<25%). Mid-branched saturated PLFA were also found at low levels; these are typical of sulfate-reducing bacteria (SRB) and Actinomycetes, which were present at all stations based on the sequencing results. Interestingly, a microeukaryotic biomarker PLFA, 18:2, was observed in surface sediments at Station 2.

**Archaeal 16S rRNA analysis**

Four different archaea phyla were detected across sediment samples including the newly designated phylum, Parvarchaeota (Fig. 3). Crenarchaeota (34%) was the most abundant phylum across all sediment samples, followed by Thaumarchaeota (15%), *Euryarchaeota* (12%) and *Parvarchaeota* (12%). Within Crenarchaeota, the majority of sequences were assigned to the classes, Deltaproteobacteria (16%) and *Gammaproteobacteria* (12%). Deltaproteobacteria dominated the upper sediment layers (0–16 cmbsf) at Station 1 and 2; the predominant order at these depths was the sulfate-reducing family, Desulfobacteriales (12%). Further, the relative abundance of sequences belonging to *Planctomycetes* and *Chloroflexi* increased with sediment depth at Station 1 and 2. The most dominant orders within *Planctomycetes* and *Chloroflexi* were *Phycisphaerae* (11%) and *Anaerolineae* (9%), respectively. The majority of sequences in surface sediments (0–4 cmbsf) at Station 3 belonged to *Gammaproteobacteria* (60%). Within *Gammaproteobacteria*, the dominant orders at this depth were *Oceanospiraliae* (18%), *Altermonadales* (13%) and *Xanthodonta*les (12%). At 4–8 cmbsf (at Station 3), *Nitrospa*ae (27%) was the most abundant phyla and a large proportion of sequences at this depth belonged to the order *Thermodesulfovibrionales* (27%), which was present in relatively low abundances at Station 1 and 2. Finally, *Planctomycetes* (42%) was the most abundant phyla at 8–12 cmbsf at Station 3 with Phycisphaerae (39%) comprising the majority of assigned sequences at this depth.

**Figure 1.** Depth distribution of total PLFA concentrations (A) and percent TOC (B) in sediment cores collected from the Caspian Sea.
Figure 2. Relative abundance of dominant bacterial groups observed in sediment cores collected from the Caspian Sea. Taxonomic distributions are depicted for the ranks of Phylum (A) and Class (B).

Figure 3. Relative abundance of dominant archaeal groups observed in sediment cores collected from the Caspian Sea. Taxonomic distributions are depicted for the ranks of Phylum (A) and Class (B).

**Statistical comparison of 16S amplicons among sediment samples**

The similarity and dissimilarity of 16S sequences across sediment samples were measured using weighted UniFrac distance metric (Fig. S3, Supporting Information). These analyses showed similar trends for bacterial and archaeal communities such that sediment samples from Station 1 and 2 showed strong clustering, while sediment samples from Station 3 had a dispersed distribution. Hierarchical cluster analysis (Fig. S4, Supporting Information) further confirmed these observations, with Station 1 and 2 forming a tight cluster, and Station 3 remaining distinct.
Information) of bacterial and archaeal communities indicated that sediment depth and sampling location may be factors contributing to variability in community structure. Accordingly, ADONIS analysis confirmed that sediment depth (R$^2$ = 0.39, P = 0.0001, strata = location) and sampling location (R$^2$ = 0.27, P = 0.0001, strata = depth) affected the observed variation among bacterial communities. Further, ADONIS analysis indicated that sediment depth (R$^2$ = 0.61, P = 0.0001, strata = location) but not sample location (R$^2$ = 0.29, P = 0.07, strata = depth) affected the observed taxonomic distribution of archaeal communities among samples.

NMDS analysis also showed that bacterial communities at Station 1 and 2 were more taxonomically similar compared to Station 3 (Fig. 4a, 2D stress: 0.065). Sediment samples within Station 3 were much more taxonomically different from one another compared to Station 1 and 2, highlighting the heterogeneity of this sediment core. Fitting of environmental variables to the bacterial NMDS ordination revealed a significant relationship (P < 0.05) between the observed pattern of taxonomic clustering with TOC (R$^2$ = 0.64, P < 0.01). This is consistent with the notion that organic carbon is one of the most fundamental factors shaping microbial communities in marine sediments (Jørgensen et al., 2012). NMDS of archaeal communities showed some clustering of sediment samples from Station 1 and 2 (Fig. 4b; 2D stress: 0.035), although more dispersion was observed. Fitting of environmental variables to the archaeal NMDS ordination found no significant correlations (P > 0.05), suggesting that the observed pattern of taxonomic clustering for archaea was less influenced by environmental factors not accounted for in this study.

**DISCUSSION**

Sediment microbial communities are vital components of marine environments and play important roles in nutrient cycling, organic matter remineralization and degradation of contaminants. Here, we combined lipid and DNA-based approaches to characterize the biomass, composition and distribution of sediment microbial communities in the Caspian Sea.

Prokaryotic cell density in marine sediments is typically 10^8 to 10^9 cells g$^{-1}$ at the surface and decreases with depth (Parkes, Cragg and Wellsbury 2000). The cell densities observed here for Caspian Sea sediments are consistent with those reported for marine sediments around the world (Parkes et al., 2000; Reed et al., 2002; Carr et al., 2013). A strong correlation between PLFA concentrations and TOC content in our study supports the notion that heterotrophy is dominant in marine sediments (D’Hondt et al., 2004; Biddle et al., 2006). PLFA concentrations were found to decrease with sediment depth; however, some increases were observed relative to the previous depths. These increases in cell density may be attributed to the increased TOC also observed at these depths. Increases in cell densities in different sediment layers have previously been observed for other marine sediments (Inagaki et al., 2003; Schippers et al., 2012; Giovannelli et al., 2013).

Microbial sulfate reduction is the major pathway for anaerobic degradation of organic matter in marine sediments.
(Jørgensen 1982; Canfield et al., 1993) and is often mediated by groups belonging to Deltaproteobacteria, specifically Desulfobacterales (Orphan et al., 2001; Leloup et al., 2009). In this study, Desulfobacterales and Desulfarculales were the dominant groups in surface and upper sediments at sampling stations with hypoxic bottom waters, Station 1 and 2 (Table S5, Supporting Information). SRB belonging to Deltaproteobacteria were present in lower abundances in surface sediments (0–4 cmbsf) at Station 3. However, the anaerobic, sulfate-reducing genus Thermodesulfovibrio (belonging to the phylum Nitrospirae) dominated sediments at Station 3 at 4–8 cmbsf, consistent with probable oxygen depletion within a few centimeters of the sediment-water interface (Jørgensen 1983; Revsbech, Madsen and Jørgensen 1986). The relative abundance of known SRB sequences across all sampling stations suggests that dissimilatory sulfate reduction may be a primary pathway for anaerobic carbon degradation in Caspian Sea sediments.

Gammaproteobacteria have been found to be one of the most abundant bacterial groups in marine sediments (Inagaki et al., 2003; Polymenakou et al., 2005; Feng et al., 2009; Mahmoudi et al., 2013b; Ruff et al., 2013). Gammaproteobacteria were present in all sediment cores in this study and accounted for a particularly large proportion of bacterial sequences in surface sediments at Station 3. Within this subphylly, Alteromonadales, Oceanospirillales and Thiotrichales were the most common orders detected. McCarren et al., (2010) found several phylogenetic groups within Alteromonadales and Thiotrichales to be stimulated with the addition high-molecular-weight (HMW) dissolved organic matter in seawater incubations indicating that these groups may play a role in the degradation of HMW-organic

Figure 5. Changes in observed OTU richness and Faith’s phylogenetic diversity (PD) with sediment depth for bacteria (A, B) and archaea (C, D).

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matter. In contrast, Oceanospirillales have been shown to aero-
ically degrade simple aliphatic hydrocarbons in marine envi-
nvironments (Hazen et al., 2010). Single-cell sequencing and meta-
transcriptomic investigations of Oceanospirillales demonstrated that members of this order have genes coding for n-alkane and cycloalkane degradation (Mason et al., 2012). Groups within Gammaproteobacteria may be playing an important role in de-
grading low- and high-MW organic matter in this system, par-
ticularly in seafloor sediments where low levels of oxygen are present in bottom waters.

Consistent with previous studies of marine sediments (Web-
ster et al., 2004; Fry et al., 2008; Blazejak and Schippers 2010), the relative abundance of Chloroflexi and Planctomycetes increased with sediment depth such that these groups were dominant in deeper sediments layers (20–36 cmbsf). Little is known about the physiology of Chloroflexi, although they are presumed to be heterotrophic (Webster et al., 2011). Members of the candidate division JS1 are key bacterial representatives associated with methane hydrates and commonly co-occur with Chloroflexi in anoxic sediment zones (Fry et al., 2008; Jørgensen et al., 2012). In this study, sequences belonging to JS1 comprised a small propor-
tion (1–5%) of the microbial community. Even though Chloroflexi and JS1 often co-occur, it has been suggested that Chloroflexi dominate organic-rich, sandy seafloor sediments while JS1 dominant strictly anaerobic, organic-rich but poor quality re-
calcitrant carbon muddy sediments with low sulfate concentra-
tions (Inagaki et al., 2006; Webster et al., 2007).

Planctomycetes have been found in high numbers (10⁶ cells mL⁻¹) in intertidal marine sediments (Musat et al., 2006) and are often detected in deep-sea and subseafood sediments (Reed et al., 2002; Inagaki et al., 2006; Harrison et al., 2009). Simi-
lar to Chloroflexi, their metabolic function in the environment is largely unexplored (Fuerst and Sagulenko 2011). The majority of Planctomycetes sequences detected in this study were assigned to the newly designated class, Phycisphaerae, which contain fac-
ultative fermentative heterotrophs (Fukunaga et al., 2009). The higher relative abundance of both Planctomycetes and Chloroflexi in deeper sediment layers indicates that they may play an im-
portant role in degrading organic matter at these deeper depths.

Sequences belonging to Thaumarchaeota accounted for a large propor-
tion of the archaeal sequences in surface sediments across all stations. Thaumarchaeota are among the most abund-
ant archaea on earth and have been detected in soils, marine waters and sediments (Francis et al., 2005; Leininger et al., 2006; Wuchter et al., 2006). Thus far, all organisms of this group have been identified as aerobic ammonia-oxidizers and possess the key enzyme, ammonia monoxygenase. Due to their ubiquity in marine environments, it is thought that Thaumarchaeota play a major role in global nitrification (Stahl and De la Torre 2012). To date, all known Thaumarchaeota have been found to be ob-
ligate aerobes; however, we detected Thaumarchaeota in sedi-
ments collected from sampling stations with hypoxic bottom waters. Similarly, Thaumarchaeota have been observed in other anoxic environments including oxygen-deficient waters (Peng, Jayakumar and Ward 2013; Parsons et al., 2014) suggesting that this group may possess alternative physiologies. Regardless, the higher prevalence of Thaumarchaeota in surface sediments ob-
served here suggests that this group may play a significant role in nitrogen cycling in the Caspian Sea.

MBGB and MCG have been shown to be ubiquitous in ma-
rine environments particularly in anoxic sediments (Inagaki et al., 2003, 2006; Biddle et al., 2006; Fry et al., 2008). A higher rel-
ative abundance of MCG to total microbes has been observed in marine sediments where sulfate penetrates at least 10 cm (Kubo et al., 2012). MCG accounted for the majority of assigned archaeal sequences at 4–8 cmbsf at Station 3; correspondingly, SRB were the most abundant bacterial group at this depth as well. In contrast, MBGB were present in fairly high abundances in all sediment samples and their relative abundance increased with sediment depth. Biddle et al., (2006) hypothesized that MBGB and MCG are anaerobic heterotrophs that consume buried carbon. More recently, it was shown that MCG play a signifi-
cant role in degrading detrital proteins in anoxic marine sedi-
ments (Lloyd et al., 2013). Thus, the prevalence of MBGB and MCG in these sediments indicates that they may also be con-
tributing to the anaerobic degradation of organic matter in the Caspian Sea.

We also detected a large number of sequences belonging to the newly assigned phyla, Parvarchaeota. Recently, phylogenetic analysis of 201 single-cell genomes enabled characterization of a new super phylum, DPANN, containing Diapherotrites, Parvar-
chaeota, Aenigmaarchaeota, Nano(halo)archaeota and Nanoarchaeota (Rinke et al., 2013). Members of this super phylum have small cell and genome sizes; little is known about their physiology or metabolism. To our knowledge, this is the first report of Par-
varchaeota in marine sediments and raises questions into their metabolic function in marine environments.

Eutrophication and resulting deoxygenation of marine wa-
ters can alter the oxidation–reduction balance in sediments as well as associated biogeochemical processes. It is difficult to discern whether sediment microbial communities in this system are directly affected by depleting oxygen levels in over-
living waters or by associated geochemical changes resulting from this depletion. Deoxygenation of overlying waters may lead to greater preservation and thus, input of organic carbon to seafood sediments. In this study, microbial biomass in surface sediments ranged from 10⁶ to 10⁷ cells g⁻¹ across sampling stations, which suggests that hypoxic bottom waters may not lead to substantial decreases in biomass of seafood microbial communities. Surface sediments collected from bottom waters that had low levels of oxygen were dominated by Gammaproteobacteria, while surface sediments collected from hypoxic bottom waters were dominated by Deltaproteobacteria, specifically SRBs. Thus, microbial sulfate reduction may become an increasingly important mechanism in seabed sediments where bottom wa-
ters become persistently hypoxic. Less evident taxonomic dif-
f erences were observed for archaeal groups in surface sediments across sampling stations, which suggests that these groups may be less sensitive to levels of oxygen in overlying water. How-
ever, it is important to note that a large portion of archaeal se-
quences were not assigned to any phyla emphasizing how little we know about archaea in marine sediments. Nevertheless, our study provides the first baseline assessment of sediment micro-
bial communities in the Caspian Sea, which could serve as the foundation for future investigations into key microbial groups and their biogeochemical role in this system.

CONCLUSION

Environmental pollution has become a significant problem in the Caspian Sea due to increased oil and gas exploration and discharge of agricultural and industrial wastewaters. Little is known about the function of even the most dominant microbial groups in the Caspian Sea, thereby limiting our understanding of the microbial metabolic potential. In this study, we demon-
strated the dominance of a number of known heterotrophs sug-
gesting that organic carbon is a primary factor shaping microbial
communities. Further, nitrogen cycling in seafloor sediments may be influenced by *Thaumarchaeota*, based on their relative abundance in surface sediments. Future studies using RNA or isotopic approaches may reveal the extent to which different groups identified in this study may be driving nutrient cycling as well as influencing the fate of contaminants.

**SUPPLEMENTARY DATA**

Supplementary data is available at FEMSEC online.

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**REFERENCES**


Revsebech NP, Madsen B, Jørgensen B. Oxygen production and consumption in sediments determined at high spatial
resolution by computer simulation of oxygen microelectrode data. Limnol Oceanogr 1986;31:293–304.


