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Effects of allochthonous DOM input on microbial composition and nitrogen cycling genes at two contrasting estuarine sites

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Running title: Effects of DOM on microbial composition and N cycling

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Summary

Heterotrophic bacteria are important drivers of nitrogen (N) cycling and the processing of dissolved organic matter (DOM). Projected increases in precipitation will potentially cause increased loads of riverine DOM to the Baltic Sea and likely affect the composition and function of bacterioplankton communities. To investigate this, the effects of riverine DOM from two different catchment areas (agricultural and forest) on natural bacterioplankton assemblages from two contrasting sites in the Baltic Sea were examined. Two microcosm experiments were carried out, where the community composition (16S rRNA gene sequencing), the composition of a suite of N cycling genes (metagenomics), and the abundance and transcription of amoA genes (quantitative PCR) were investigated. The river water treatments evoked a significant response in bacterial growth, but effects on overall community composition and on the representation of a suite of N cycling genes were limited. Instead, treatment effects were reflected in the prevalence of specific taxonomic families, specific N related functions, and in the transcription of amoA genes. The study suggests that bacterioplankton responses to changes in the DOM pool are constrained to part of the bacterial community, whereas most taxa remain relatively unaffected.
Introduction

Marine heterotrophic bacterioplankton process dissolved organic matter (DOM), thereby mineralizing nutrients essential for growth of phytoplankton and affecting overall productivity in marine waters (Azam et al., 1983). Among nutrients, nitrogen (N) is a primary constituent of various cellular macromolecules, and the availability of N is commonly a limiting factor for primary and secondary production in diverse marine systems (Ryther and Dunstan, 1971; Bristow et al., 2017). In marine coastal systems, the release of dissolved inorganic N (DIN) through the degradation of dissolved organic nitrogen (DON) can be orders of magnitude higher than the input of DIN from land (Knudsen-Leerbeck et al., 2017). Hence, N released or acquired through microbial degradation of nitrogenous DOM is an important N source for bacterioplankton and phytoplankton growth (Bronk et al., 2007).

Bacterioplankton control not only the accessibility of N through DOM processing, but also regulate the oxidative state of N present in the environment through a series of oxidative and reductive processes. Consortia of microorganisms mediate key steps in the marine nitrogen cycle (Falkowski et al., 2008), including e.g. ammonification, nitrogen fixation, nitrification, and denitrification (Zehr and Ward, 2002), and ultimately determine the availability of N for higher trophic levels, e.g. phytoplankton. For instance, the form and oxidation level, e.g. whether inorganic N is available as ammonia or nitrate, may affect both the productivity and the composition of the phytoplankton community (Glibert et al., 2016).

In estuarine environments, riverine DOM is an important source of highly labile N (Seitzinger et al., 2005; Bronk et al., 2007). The characteristics of the riverine DOM may depend on the catchment area and on season. Consequently, it is conceivable that the riverine input,
particularly in N limited environments, selects for bacterioplankton capable of hydrolyzing
nitrogenous DOM and for taxa involved in down-stream nitrogen cycling processes. Moreover, the
bacterial community response would likely rely on the availability and nature of the nitrogenous
DOM and depend on bacterioplankton community composition and contemporary environmental
conditions. Hence, responses are expected to differ between localities. While these assumptions
appear logical, they have to our knowledge not been experimentally verified.

The Baltic Sea is the second largest estuarine system in the world and encompasses
separate sub-basins with unique geology and a strong north-south salinity gradient driven by river
outlets (Rönnberg and Bonsdorff, 2004). The north is characterized by high DOM concentrations
and phosphorous (P) limited plankton production whereas the south has lower DOM
concentrations and N limited plankton productivity (Bernes, 2005; Rowe et al., 2018). Further,
catchment characteristics vary from primarily forest in the north to agricultural landscapes in the
south. The gradient in biogeochemistry is also reflected in extensive changes in microbial
community composition from north to south (Herlemann et al., 2011). Climate change is predicted
to increase precipitation and the allochthonous DOM input via rivers to the Baltic Sea (Andersson
et al., 2015). The loading and characteristics of the DOM will likely affect the future microbial
community composition, activity, DOM utilization, and nutrient biogeochemistry in the Baltic Sea
(Traving et al., 2017; Rowe et al., 2018). However, responses will conceivably vary between north
and south due to differences in catchments, characteristics of the incoming DOM, and
composition of the recipient microbial communities.

In the present study we examined effects of riverine DOM loading in incubation
experiments in two contrasting environments; the southern Baltic Sea (Øresund) after the spring
bloom and in the northern Baltic Sea (Storfjärden) in summer (Fig. S1). After the spring bloom, Øresund surface water is typically N depleted whereas the surface water at Storfjärden typically has higher N and DOM concentrations during the summer. We tested the effects of allochthonous DOM loads at both sites by additions of river water from an agricultural and a forest (humic) catchment area, and examined the microbial community composition (16S rRNA genes), the abundance of nitrogen cycling genes (reflecting the metabolic capacities) and the activity of ammonia oxidizers. Since effects on community composition and the composition of functional genes may not be detectable during short-time incubations, we chose to examine changes in functional gene transcription (as a proxy for activity), namely, quantifying the transcription of amoA genes (coding for ammonia monooxygenase) - genes involved in nitrification - which is a critical N cycling process facilitating N loss through coupled nitrification-denitrification in the Baltic Sea coastal zone (Hietanen et al., 2012). We anticipated temporal functional succession and treatment-specific responses to our manipulations, and further that effects would differ between the two localities with a more modest response in the relatively DOM and nitrogen-rich northern locality.

Experimental procedures

**Experimental setup for Exp I and Exp II**

The setup described here was part of a larger experiment reported elsewhere (Markussen et al., 2018). Microcosm experiments with the same setup were conducted at the Marine Biological Laboratory (University of Copenhagen, Denmark) using Øresund water (Exp I) and at the Tvärminne Zoological Field station (University of Helsinki, Finland) using water from Storfjärden,
Gulf of Finland (Exp II). The microcosms consisted of three treatments in triplicates (control in 6 replicates) including control, addition of water from a humic river (hereafter DOMhum) and an agricultural river (DOMagri). Water was collected on April 20th 2015, Øresund (56°3’ 26.4” N, 12°38’44.9” E) from 5 m (300 L) and 12 m (10 L) depth, and on July 27th 2015, at Storfjärden, Gulf of Finland (59°51’11.9”N 23°16’19.2”E) 300 L from 0, 5, and 10 m depth (Fig. S1). The water was prefILTERED through a 10 m plankton net, filtered through a 0.22 m capsule filter (Optical XL, Millipore), pooled to ensure homogeneity, and filled into microcosms (10 L PC bottles, Nalgene) to represent 60% of the final volume, except for the controls that were filled to represent 80% final volume. The river water was collected 2 days prior to the start of the experiments from the rivers Lapväarti (62°14’20.6”N 21°34’37.5”E, Finland) and Lielupe (56°48’41.6”N 23°35’04.9”E, Latvia; Fig. S1), filtered through a 0.22 m capsule filter (Optical XL, Millipore), and stored at 4°C until use. The salinity of the river water was adjusted with muffled NaCl to in situ levels of 13.4 (Øresund) and 6 (Storfjärden) prior to addition to the microcosms. River water was added to the microcosms representing 20% of the final volume. A plankton inoculum (<10 μm) was added to each batch, representing 20% of the final volume, to initiate the experiments. The microcosms were incubated in the dark, in a temperature-controlled room at in situ temperature ±3°C. Exp I and Exp II had a duration of 5 and 4 days, respectively. Daily samplings at 09.00 and 21.00 covered a variety of environmental parameters and DNA/RNA sampling.

**Bacterial abundance and production, and nutrients**

Data were adapted from Markussen et al. (2018). Briefly, Samples for bacterial enumeration were fixed with glutaraldehyde (1% final conc., Sigma-Aldrich), stored at -80°C, and later enumerated using SYBR green I (Invitrogen) and a BD FACS Canto II flow cytometer. Bacterial
production was measured by $^3$H-thymidine incorporation (Fuhrman and Azam, 1982). Ammonium ($\text{NH}_4^+$) concentrations were determined directly from fresh samples using ortho-phthalaldehyde (Holmes et al., 1999) and a rapid flow analyser (Turner Designs Trilogy Laboratory Fluorometer). Nitrate ($\text{NO}_3^-$) and phosphate ($\text{PO}_4^{3-}$) were measured on an auto-analyser according to (Wood et al., 2009) and (Murphy and Riley, 1962), respectively. Dissolved organic carbon (DOC) and nitrogen (DON) was measured on a Shimadzu TOC-L Total Organic Carbon Analyzer (Shimadzu Corporation) as previous described (Paulsen et al., 2017).

**DNA and RNA sampling and extraction**

Water was sampled in situ, from the inoculum, and twice daily from each microcosm during the experiments. One liter water was collected in 1 L PC bottles (Nalgene), immediately mixed with 100 ml stop-solution (5% phenol in 99.8% ethanol (Khodursky et al., 2003)), and stored for < 24 h at RT in the dark. Fixed samples were then filtered onto 0.22 µm membrane filters (Durapore GVWP04700, Milipore) and stored at -80°C. Nucleic acids were extracted from the filters using the Allprep kit (Qiagen), and then purified and concentrated using the RNA Clean & Concentrator (Zymo) and Genomic DNA Clean & Concentrator (Zymo). DNA and RNA extracts were quantified using Quant-IT RiboGreen and PicoGreen (Invitrogen), respectively.

**16S rRNA gene amplicon sequencing and metagenomics**

For determining the community composition 16S rRNA genes were amplified from total DNA using the primers Bakt_341F (CCTACGGGNGGCWGA) and Bakt_805R (GACTACHVGGTATCTAATCC) (Herlemann et al., 2011), and products were sequenced with on an Illumina MiSeq paired-end multiplex platform at SciLifeLab/NGI (Solna, Sweden). Raw amplicon
reads were quality trimmed (Trimmomatic ver 0.32), and chimeras removed and reads assigned
into operational taxonomic units (OTUs) using 97% cut-off in cd-hit-otu (Li et al., 2012). SINA 1.2
(Pruesse et al., 2012) was used against the SILVA database (v. 115) to classify unique OTUs and the
relative abundance of each OTU was estimated using an in-house Python script. Sequences were
deposited in NCBI SRA (Bioproject number 542 PRJNA435478).

DNA (2-10 ng) from each sample was prepared for metagenomics sequencing with
the Rubicon ThruPlex kit (Rubicon Genomics, Ann Arbor, Michigan, USA) according to the
instructions of the manufacturer. Cleaning steps were performed with MyOne carboxylic acid-
coated superparamagnetic beads (Invitrogen, Carlsbad, CA, USA). Libraries were sequenced on a
HiSeq 2500 (Illumina Inc., San Diego, CA, USA). On average, 18 million paired-end reads (125 bp)
per sample were generated. Raw reads were quality trimmed with Cutadapt (Martin, 2011) from
both read ends and duplicate reads were removed with fastuniq (Xu et al., 2012). High quality
reads were mapped on the BARM database containing the most informative reference genomes in
the Baltic Sea (Alneberg et al., 2018) with Bowtie2 using default parameters (Langmead et al.,
2013). The raw counts were calculated from Bedtools histogram output (Quinlan, 2014) and
quantitative abundance of reads were annotated using Clusters of Orthologous Groups (COG)
(Galperin et al., 2015).

Reverse transcription and quantification of amoA genes and transcripts
cDNA was synthesized using gene-specific reverse primers and the TaqMan Reverse
transcription kit (Life Technologies) according to manufacturer’s protocol. The RT products from
each sample were quantified using PicoGreen and used to calculate the efficiency of the RT
reactions (RT factor; conversion factor of RNA to cDNA). PCR amplification using universal 16S rRNA gene primers was tested on RNA extracts to confirm complete DNAse digestion.

AmoA genes and gene transcripts from ammonia oxidizing archaea (AOA) and bacteria (AOB) (β-proteobacteria) were quantified from extracted DNA and RNA (cDNA) according to Happel et al. (2018) using a BioRad ddPCR system and the primer sets Arch-amoAF (5'-STAATGGTCTGGCTTAGACG-3') and Arch-amoAR (5'-GCGGCCATCCATCTGTATGT) (Francis et al., 2005), and amoA-1F (5'-GGGGTTTCTACTGGTGGT-3') and amoA-2R (5'-CCCCTCKGSAAAGCCTTTC-3') (Rotthauwe et al., 1997), respectively. Each reaction mixture (25 µl) consisted of 10 µl Evagreen ddPCR mix (Bio-Rad), 200 nM of each primer, BSA (0.5 g µl⁻¹) and ca. 20 ng of template. The mixture was loaded with 70 µl Evagreen droplet generation oil into a droplet generator. PCR was performed in a T100 thermal cycler using a profile of 95°C for 10 min, followed by 40 cycles of 94°C for 30 s and 60°C (AOA) or 53°C (AOB) for 60 s, 1 cycle of 98°C for 10 min, and ending at 4°C. Droplets were read on the droplet reader and data analyzed using the QuantaSoft software (Bio-Rad). Quantification was presented as the number of target molecules per µl of PCR mixture and converted to copy number per liter seawater using the volume of water filtered. RNA sample quantifications were corrected using the RT factor of each sample. Equal extraction efficiencies for all samples are assumed.

Statistical analysis

To compare the bacterial communities, the taxonomic richness (calculated as abundance-based coverage estimator (ACE)) and the Shannon diversity index were estimated (Markussen et al., 2018). Significant differences (P<0.05) between bacterial abundance and
production data were tested using Dunnett’s test (Markussen et al., 2018). Raw counts of EC/PFAM/COGs were normalized to counts per million (cpm) using EdgeR (Robinson et al., 2010) to test for significant differences in COG abundances between control samples and treatments. Only COGs with an FDR <0.05 and p<0.005 were considered significant. Generalized linear models (GLM) were investigated to test for significant correlations between the relative abundances of EC/COG/PFAMs and environmental parameters using the mvabund (Wang et al., 2012) package in R. 16S rRNA gene OTU count data were normalized using DESeq2 (Love et al., 2014) and principal component analysis (PCA) was done using the R package. ANOSIM was done in R to test if community composition differed between sites. ANOVA was used to test if amoA gene and transcript abundances differed between treatments. Pearson’s product moment coefficients were used to test for correlations between environmental parameters and amoA gene and transcript abundances.

Results and discussion

The effect of agricultural and humic river water DOM on microbial communities

In both experiments, there was a general increase in concentrations of DOC, DON, ammonium (NH₄⁺) and nitrate (NO₃⁻) in the DOMhum and DOMagri treatments relative to the controls. In particular high DON (104 μM) and NO₃ (62 μM) concentrations were found in the DOMagri treatment in Exp I, whereas both river treatments had high DOC concentrations (711 and 756 μM, respectively; Table 1) in Exp II.

Bacterial production (BP) (Fig. 1A, C) and abundance (BA) (Fig. 1B, D) increased significantly over time in both experiments, but with large differences between the experiments.
BA (both experiments) and BP (Exp II) were significantly increased in both the DOMagri and
DOMhum treatments compared with the controls, while BP was not significantly stimulated by
either of the treatments in Exp I. The observed growth responses were anticipated to be
accompanied by community dynamics mirrored in composition (16S rRNA genes) and function
(composition and transcription of N cycling genes), based on earlier studies reporting that DOM
can shape bacterioplankton community composition (McCarren et al., 2010; Landa et al., 2015;
Traving et al., 2017). However, a PCA revealed that community composition at the end of the
experiments clustered into site rather than treatment (Fig. 2A). Moreover, the community
composition differed significantly between Exp I and II (ANOSIM, $r^2=0.44$, $p<0.001$) but not
between treatments (ANOSIM, Exp I+II: $r^2=0.04$, $p=0.88$, Exp I: $r^2=0.10$, $p=0.69$, Exp II: $r^2=0.49$,
$p=0.09$). Similarly, Shannon diversity ($r^2 = 0.67$, $p < 0.001$) and taxonomic richness was significantly
higher in Exp II compared to Exp I ($r^2 = 0.24$, $p < 0.001$); however, no significant differences in
alpha-diversity were observed between treatments from the same experiment. Hence, the
changes in bacterial growth were only accompanied by limited shifts in community composition –
and this was observed in both examined environments with marked differences in local
community composition.

At the phylum level, e.g. γ-proteobacteria were significantly over-represented in the
DOMagri treatment in Exp I and in both DOMagri and DOMhum in Exp II (Fig. 3A). In Exp II, e.g. β-
proteobacteria were stimulated in the DOMhum treatment relative to the controls. Such
stimulation of β-proteobacteria by DOM has previously been observed for Baltic bacterioplankton
(Kisand and Wikner, 2003; Traving et al., 2017). At the family level, there were several responses
within Proteobacteria (Fig. 3B); e.g. Alteromonadaceae was more abundant in the DOMagri
treatment relative to control in Exp I (DOMagri: 58%; Control: 25%). Within one of the abundant
groups, Bacteroidetes, responses were limited (Fig. 3C). Hence, some differences were observed in composition between treatments (Fig. 3), but overall changes were considerably less than the difference between environments (Fig. 2). There are examples of resistant microbial composition withstanding disturbance (e.g. Bowen et al., 2011); however, it may also be that DOC manipulations, as in the current study, only select for some taxa whereas the relative abundance of most taxa remain unchanged. Hence, it appears that overall community structure is a relatively poor predictor of the bacterial growth response, as also suggested by others (Dinasquet et al., 2013).

Relative abundance of nitrogen cycling genes

It has been suggested that the key level at which to address the assembly and structure of bacterial communities is not taxonomy but rather the more functional level of genes (Burke et al., 2011; Krause et al., 2014). Moreover, since N availability affects N cycling genes (e.g. Zhang et al., 2013), we hypothesized that the high N concentrations in the added river water would elicit extensive and differential responses in the relative abundance of N cycling genes at the two sites. Nevertheless, as in the compositional analysis, a PCA of the relative abundance of N cycling genes (EC/EggNOG/PFAM; see Table S1) revealed a clustering according to site (ANOSIM, $r^2=0.1944$, $P=0.003$) rather than treatment (ANOSIM, Exp I+II: $r^2=0.06$, $p=0.62$, Exp I: $r^2=0.15$, $p=0.50$, Exp II: $r^2=0.51$, $p=0.037$) (Fig. 2B). Generalized linear models (GLM) showed that the relative abundances of all N cycling genes did not correlate with any environmental parameters in Exp I. In Exp. II, on the other hand, there were significant correlations with NH$_4$ (LR=1438.8, $p=0.026$), DOC (LR=1592.3, $p=0.011$), DON (LR=1508.8, $p=0.012$) and treatment (LR=2665.3, $p=0.018$). This suggests that while initial natural communities had a significant impact on the functional response
to the river water amendments, addition of river water with a high DOC to DON ratio in Exp. II (Table 1) also affected the abundance of N cycling genes.

Despite that the community analysis of N cycling genes did not reveal a clustering according to treatment (Fig. 2B), the relative abundance of some specific genes did differ significantly between controls and treatments (see below). However, in line with the above GLM results, more were over- or under-represented in Exp II than Exp I (Fig. 4). This suggests that a universal response (across sites) in N cycling genes due to treatment alone was not the case, but rather that the community of the northern Baltic Sea (Exp II; Storfjärden) was more responsive than that of the southern Baltic Sea (Exp I; Øresund). Reasons for this may include multiple site characteristics or seasonality (sampling in April vs. July); however, it is noteworthy that DOC levels naturally, and in our experiment, are highest in the northern Baltic (Table 1) (Sandberg et al., 2004; Rowe et al., 2018). Bacterioplankton in this environment may, therefore, be particularly adapted and responsive to pulses of riverine DOM. In addition, the higher diversity and taxonomic richness of Storfjärden could possibly have benefited the responsiveness of this community.

In Exp I, a N$_2$ fixation related gene (nifB, COG0535) and a nitrous oxide reduction gene (nosZ, PF13473) were over-represented in the DOMhum treatment relative to the control. Further, both ammonia transporters (COG0004, PF00909) and nitrite/nitrate reductases (PF07732, PF00394, PF04879) were under-represented (Fig. 4A). The DOMagri treatment did not have any significant effect on the relative abundance of N related genes (Fig. 4B). In Exp II, several EC/COG/PFAMs (DOMhum: 20; DOMagri: 13) differed significantly in relative abundance between treatments and controls (Fig. 4C,D). Among these, some of the N$_2$ fixation related genes were significantly over-represented in the DOMhum treatment (COG1348, PF00142, PF00148, EC...
1.18.6.1) while one was under-represented (PF04055) (Fig. 4C). Most ammonia and nitrite/nitrate transporters were under-represented (PF00909, COG0004, PF07690) along with two urease genes (PF07969, PF01979). Both nitrate reductase (EC 1.7.99.4) and nitrous oxide reductases (COG4263, EC 1.7.2.4) were over-represented. In the DOMagri treatment, a single $N_2$ fixation gene was under-represented (COG0535) while both nitrite/nitrate transporters (COG2223) and nitrous oxide reductases (COG4263, EC 1.7.2.4, EC 1.7.1.14) were over-represented (Fig. 4D).

While there were only few responses in N related genes to the DOMagri treatments, there were several overlaps in the response to the DOMhum treatment between the two experiments. The under-representation of ammonia channel protein AmtB and over-representation of $N_2$ fixation genes in the DOMhum treatments, both point to possible N limitation (Carini et al., 2018). There were, however, no indications of N limitation when looking at the N:P ratios. The N:P ratios (calculated as $(NH_4+NO_3)/PO_4$) were highest in the DOMagri treatments in Exp. I and in the DOMhum treatment in Exp. II. Further, the over-representations of nitrate reductases (Exp I) and nitrous oxide reductases (both experiments) could indicate a promotion of some steps of the denitrification pathway by the addition of DOMhum.

Denitrification in the Baltic Sea is known from anoxic zones of the water column (Dalsgaard et al., 2013) and from sediments (Silvennoinen et al., 2007).

**Abundance and activity of ammonia oxidizing archaea (AOA) and bacteria (AOB)**

To quantitatively assess the impact of the treatments on functional gene abundance and transcription, digital droplet PCR (ddPCR) was used to enumerate *amoA* genes and transcripts of AOA and AOB in each experiment initially, after 44-45 h, and at the end of each experiment (Fig. 5). The ddPCR method was chosen because of its documented ability to quantify *amoA* genes from
Baltic Sea waters (Happel et al. 2018). For principles and details of the ddPCR methodology, please see the recent study by Happel and co-authors (2018).

Although amoA gene and transcript abundances of both AOA and AOB were dynamic over time, there was no significant treatment effect on amoA abundances. There were, however, significant differences between the amoA transcript abundances of DOMhum and control treatments in Exp I for both AOA (one-way ANOVA; F=11.7, p=0.011 and one-way ANOVA on ranks; Q=2.6, p=0.014) and AOB (one-way ANOVA on ranks; Q=2.32, p=0.024 and Q=2.55, P=0.032, respectively) at the beginning of the experiment. In both experiments, we found abundances of AOA (3.9 x 10³ – 7.3 x 10⁴ copies L⁻¹) and AOB (4.2 x 10³ – 5.4 x 10⁴ copies L⁻¹) in similar ranges, whereas amoA gene transcription was dominated by AOB (up to 1.5 x 10⁹ copies L⁻¹ for AOB and 1.5 x 10⁸ copies L⁻¹ for AOA). Concentrations of AOA and AOB were remarkably low compared to coastal environments (Beman et al., 2008, Hollibaugh et al., 2011), but similar to the Yangtze River estuary (Zhang et al., 2014) and to our previous study on ammonia oxidizers in the south and north of the Baltic Sea (Happel et al., 2018). Moreover, the dominance of amoA transcripts from AOB also matches our previous Baltic Sea study (Happel et al., 2018). Surprisingly, there was no significant stimulation of amoA gene abundances from AOA or AOB by the addition of riverine DOM in either experiment and amoA gene transcription was in fact reduced in the DOMhum treatments relative to controls. This could be interpreted as a sign of ammonia limitation (Carini et al., 2018), however, since amoA gene transcription from AOB was negatively correlated with DOC in Exp I (Pearson correlation, r=-0.34, p=0.04), we speculate that ammonia oxidizers were hampered by the introduction of riverine DOC – potentially the sudden availability of labile riverine carbon is at odds with the chemolithoautotrophic life strategy of ammonia oxidizers (Strauss and Lamberti, 2000; Strauss et al., 2002).
Concluding remarks

Despite that the addition of river water caused several folds increase in bacterial growth in both the Øresund and Storfjärden experiments, only specific sub-populations and N cycling processes were affected by the treatments, whereas overall community composition and the collective pool of examined N cycling genes remained relatively unaffected. This may support the notion that many bacterioplankton species are generalists and less responsive to transient changes in the DOC pool (Mou et al., 2008), and that the linkage between identity and specialized DOC utilization is valid only within some phyla or among specific sub-populations (Dinasquet et al., 2013).

Interestingly, treatment effects on nitrification were only observed at the transcription level, and not the gene level. This observation underlines that functional responses in key N cycling processes in bacterioplankton may not always be accompanied by selection affecting community composition. Our study included several experimental variables like seasons and river inocula which prevents firm comparisons between the two localities; south and north in the Baltic Sea. Nevertheless, the higher responsiveness of the community in Storfjärden to riverine DOM sources is noteworthy. If coupled with the projected future increases in precipitation and outlet of allochthonous DOM (Andersson et al., 2015), we speculate that the coastal zones in the Northern Baltic Sea will undergo more dramatic future changes in N cycling regimes than the communities in the Øresund. However, focused studies are needed in order to validate this hypothesis.

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References


Figure 1. Bacterial abundance and bacterial production during Exp I (Øresund) (A, B) and Exp II (Storfjärden) (C, D) for the controls and the agriculture (DOMagri) and the humic (DOMhum) river water treatments. Data are adapted from Markussen et al. (2018). Error bars indicated SD of biological triplicates (6 replicates in control samples).
Figure 2. Principal component analysis (PCA) of community composition (based on 16S rRNA sequencing; A) and nitrogen related genes (EC/eggNOG/PFAM) (metagenomics sequencing; B) at the end of Exp I (Øresund) and Exp II (Storfjärden) for the controls and the agriculture (DOMagri) and humic (DOMhum) river water treatments. For separate PCA of each experiment; see Figure S2.
Figure 3. Relative abundance of phyla/sub-phyla (A), families within Proteobacteria (B) and Bacteriodetes (C) at the end of Exp I (Øresund) and II (Storfjärden) for the controls and the agriculture river water (DOMagri) and the humic river water (DOMhum) treatments. Significant differential abundances for each group between treatments were tested using EdgeR and indicated for Exp I (*) and Exp II (°).
Figure 4. Volcano plots showing up- and down-represented EC/PFAM/COGs in Exp I (A, B) and Exp II (C, D) for the controls and the humic (DOMhum)(A, C) and the agriculture (DOMagri) (B, D) river water treatments. Significantly differentially abundant EC/PFAM/COGs are marked with black dots.
Figure 5. Archaeal (AOA) and bacterial (AOB) amoA gene and transcript abundances at the start of the experiments (A,D), after 44-45 hours (B,E) and at the end of the experiments (C,F) for Exp I (left) and Exp II (right) for the controls and the agriculture (DOMagri) and humic (DOMhum) river water treatments. Note the different scales in (B). Error bars indicated SD of biological triplicates (6 replicates for control samples).
Table 1. Concentrations of dissolved organic carbon (DOC), dissolved organic nitrogen (DON) ammonium (NH$_4$), phosphate (PO$_4$) and nitrate (NO$_3$) in the treatments at the beginning of the experiments for the control, the agriculture river water treatment (DOMagri) and the humic river water treatment (DOMhum). Standard deviations in brackets.

<table>
<thead>
<tr>
<th>Treatment</th>
<th>Exp I (Øresund, Southern Baltic Sea)</th>
<th>Exp II (Storfjärden, Northern Baltic Sea)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Control</td>
<td>DOMhum</td>
</tr>
<tr>
<td>DOC (µM)</td>
<td>381.50 (13.66)</td>
<td>552.67 (51.43)</td>
</tr>
<tr>
<td>DON (µM)</td>
<td>47.37 (0.37)</td>
<td>51.97 (1.34)</td>
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<td>NH$_4$ (µM)</td>
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<td>0.94 (0.12)</td>
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<tr>
<td>PO$_4$ (µM)</td>
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<td>0.20 (0.02)</td>
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<tr>
<td>NO$_3$ (µM)</td>
<td>1.74 (0.19)</td>
<td>3.63 (0.66)</td>
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</tbody>
</table>