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Molecular insights into the microbial formation of marine dissolved organic matter: recalcitrant or labile?

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Abstract

The degradation of marine dissolved organic matter (DOM) is an important control variable in the global carbon cycle and dependent on the DOM composition. For our understanding of the kinetics of organic matter cycling in the ocean, it is therefore crucial to achieve a mechanistic and molecular understanding of its transformation processes. A long-term microbial experiment was performed to follow the production of non-labile DOM by marine bacteria. Two different glucose concentrations and dissolved algal exudates were used as substrates. We monitored the bacterial abundance, concentrations of dissolved and particulate organic carbon (DOC, POC), nutrients, amino acids, and transparent exopolymer particles (TEP) for two years. Ultrahigh resolution Fourier Transform Ion Cyclotron Resonance Mass Spectrometry (FT-ICR-MS) allowed the molecular characterization of extracted DOM after 70 days and after ~ 2 years of incubation. Although glucose was quickly degraded, a DOC background was generated in glucose incubations. Only 20 % of the organic carbon from algal exudate was degraded within the 2 years of incubation. TEP, which are released by micro-organisms, were produced during glucose degradation but decreased within less than three weeks back to half of the maximum concentration and were below detection in all treatments after 2 years. The molecular analysis demonstrated that DOM generated during glucose degradation differed appreciably from DOM produced during the degradation of the algal exudates. Our results led to several conclusions: (i) Higher substrate levels result in a higher level of non-labile DOC which is an important prerequisite for carbon sequestration in the ocean; (ii) TEP are generated by bacteria but are also degraded rapidly, thus limiting their potential contribution to carbon sequestration; (iii) The molecular signatures of DOM derived from algal exudates or glucose after 70 days of incubation differed strongly from refractory DOM. After 2 years, however, the molecular patterns of DOM in glucose incubations were more similar to deep ocean DOM whereas the degraded exudate was still different.

BGD
11, 3065–3111, 2014

1 Introduction

Refractory dissolved organic matter (DOM) in the oceans represents a large reservoir of organic carbon in the global carbon cycle (642 Pg C, Hansell, 2013). The ultimate sources of marine DOM are primary production in the sun-lit surface layer of the ocean, and continental runoff. If newly generated organic carbon is converted into refractory substances, it becomes an important sink for atmospheric carbon on medium-term time scales (Carlson et al., 1994, 2010). An efficient sequestration of DOC (86 $Tg\,C\,yr^{-1}$, Hansell et al., 2009) occurs in areas of deep-water formation, particularly in the polar oceans. However, this process requires long DOM residence times to survive a downward transport and the cycling within the deep ocean currents.

The average dissolved organic carbon (DOC) atom in the ocean has a radiocarbon age of 4000–6000 years (Bauer et al., 1992) but for specific DOM fractions or molecules the residence time can be substantially longer (Loh et al., 2004; Lechtenfeld et al., 2014). It is not clear how this potential carbon buffer will evolve in the future global biogeochemical cycle and how it effects the climate system (Denman et al., 2007). The efficacy of forming recalcitrant organic matter out of labile substrate, which depends on the environmental conditions, is critical for the magnitude of the sequestration flux.

Microbial utilisation and modification is probably the most important process for the formation of refractory DOM (Jiao et al., 2010 and references therein). In experimental incubations marine bacteria form non-labile organic matter from simple substrates such as glucose. These non-labile substances persist for up to more than a year (e.g. Skoog et al., 1999; Ogawa, 2001; Gruber et al., 2006), suggesting that microbial activity may indeed be responsible for converting labile photosynthates into refractory organic matter. It is unknown, however, whether the organic material produced in such experiments is chemically similar to refractory DOM and therefore could be preserved on time scales beyond those achievable in lab incubations.

The persistence of refractory DOC in the ocean is attributed to its intrinsic chemical stability (e.g. Koch et al., 2005; Hertkorn et al., 2006) and its low concentration,

BGD
11, 3065–3111, 2014

especially in the deep ocean (Kattner et al., 2011). Since pelagic microbial communities can differ widely in their abilities to express specific extracellular enzymes, their composition can also decide on the degradability of a substrate (e.g. Arnosti, 2004; Carlson et al., 2004). The lack of essential inorganic nutrients and bioavailable organic substrates (called co-metabolism or priming effect) can also impede microbial degradation of organic matter (e.g. Horvath, 1972; Alderkamp et al., 2007; Bianchi, 2011). It is likely that a combination of all of these factors leads to the long average turnover time of marine DOM.

A surface active fraction of marine DOM, which is rich in acidic polysaccharides, forms a class of particles called Transparent Exopolymer Particles (TEP) (Mopper et al., 1995; Zhou et al., 1998; Passow, 2000; Verdugo and Santschi, 2010). Exudates from phytoplankton and bacteria are often rich in TEP and their dissolved precursors (Myklestad, 1995; Passow, 2002b; Ortega-Retuerta et al., 2009), which exist in a size continuum, from fibrillar macromolecules (Leppard, 1995) released by many organisms to TEP 100's of micrometres long (Verdugo et al., 2004). As particulates, TEP, play an essential role for aggregation and vertical flux of particulate organic matter (POM) (Alldredge et al., 1993; Passow, 1994; Logan et al., 1995). It has even been suggested that TEP concentration determines aggregation (Passow, 1994; Arrigo, 2007; Gardes et al., 2011). TEP also provide surfaces and substrate for bacteria and archeae, creating hot spots of microbial activity (Smith et al., 1992; Passow and Alldredge, 1994; Azam and Long, 2001). Despite their dominant role in marine carbon cycling, very little is known about the lability of TEP, especially on timescales longer than one or two weeks (Passow, 2002a).

In the past, in vitro degradation experiments mainly provided bulk chemical characteristics and molecular information for a small fraction of refractory DOM (e.g. Lara and Thomas, 1995; Ogawa et al., 2001). The application of ultrahigh-resolution Fourier transform ion cyclotron resonance mass spectrometry (FT-ICR-MS) resulted in major advances in the molecular characterization of complex organic matter samples (e.g. Kujawinski et al., 2002; Stenson et al., 2002). The technique is suitable to identify

BGD
11, 3065–3111, 2014

molecular fingerprints of different sources (e.g. Hughey et al., 2007; Gonsior et al., 2011; Schmidt et al., 2011) and transformation processes (Rodgers et al., 2000; Kujawinski et al., 2004) of organic matter. Based on FT-ICR-MS analyses it has been previously hypothesized that the molecular composition of all refractory organic matter is similar and independent of its ultimate source (Koch et al., 2005; Rossel et al., 2013). Conceptually, this contradicts the findings that sources and transformations processes are molecularly imprinted in organic matter. A better mechanistic understanding of the kinetics which convert labile material (which contains the original biochemical signal) into refractory organic matter (which represents the sequestration potential) is required to resolve this contradiction. So far, such kinetics are not well constrained, although crucial for the conservation of molecular biomarker signals.

In our experiments we investigated changes in DOM and TEP concentrations and shifts in the molecular composition of DOM during the microbial utilization of glucose or an algae exudate during a period of 2 years. The main goals of the study were to test the hypothesis that the degradation of different substrates leads to refractory DOM with similar molecular characteristics. Specifically, we tested if DOM with refractory molecular characteristics can be generated on time scales of less than three months and if organic nitrogen is incorporated into persistent DOM. We also examined if the addition of labile substrates results in an increased mineralization of refractory DOM (co-metabolism; priming effect) and verified the idea that the majority of TEP are labile and removed within weeks.

2 Material and methods

2.1 Experimental setup

Eleven 50 L glass bottles containing seven treatments and four controls were incubated for 70, 695 or 734 days in the dark at 0 °C (Table 1). The general design of the exper-

BGD
11, 3065–3111, 2014

iment followed that of Ogawa et al. (2001). Briefly, substrate and bacterial inoculum were added to sterile artificial seawater and changes in DOM were monitored.

The incubation bottles (Table 1) consisted of (i) three replicate treatments that contained glucose ([Glu]), (ii) two that contained dissolved algae exudates ([exud]) and (iii) two that contained ^{13}C-labeled glucose ([^{13}Glu]; D-Glucose-1-^{13}C, Sigma). These treatments were inoculated with bacteria. Additionally, four different controls were prepared: the two background controls received inoculum or sterilely filtered inoculum, respectively, but no substrate (^{c}[none]) and (^{sc}[none]). The two other sterile controls received sterilely filtered inoculum and either exudates (^{sc}[exud]) or glucose (^{sc}[Glu]). A natural microbial community collected from Antarctic water was used as the bacterial inoculum. After 699 days of incubation, glucose was added to one [exud] and one [Glu] treatment to evaluate the potential influence of co-metabolism on DOC degradation (Table 1). Samples for bulk parameters were collected at 11 to 15 time steps during the incubations and samples for the DOM extraction and molecular characterization by ultrahigh resolution mass spectrometry were collected after 70 and 695 days.

Results are primarily presented as averages of replicates with identical substrates ($n = 3$ for glucose, $n = 2$ for DOM, $n = 2$ for ^{13}C labeled glucose). In the following, the treatments are labeled with the type of substrate in square brackets ($[\mathrm{Glu}]_x$; $[\mathrm{exud}]_x$; or $[^{13}\mathrm{Glu}]_x$). The subscript index (x) indicates the day or period of sampling if applicable.

2.2 Preparation of experiment

The 50 L glass bottles were acid (HCl, 2 M, Merck, p.a.) and base washed (NaOH, 2 M, Merck, p.a.) and then rinsed with ultrapure water (Millipore). Each bottle was filled with ~ 45 L of sterile filtered (0.2, Polycap, Whatman) artificial seawater, containing NaCl (24.99 g L^{-1}), $MgCl_2 \cdot 6H_2O$ (11.13 g L^{-1}), Na_2SO_4 (4.16 g L^{-1}), $CaCl_2 \cdot 2H_2O$ (1.58 g L^{-1}), KCl (0.79 g L^{-1}), and $NaHCO_3$ (0.17 g L^{-1}) dissolved in ultrapure water. All salts except $CaCl_2$ (sterile filtered) were pre-combusted (500 °C, 5 h) before use. Sterile

BGD
11, 3065–3111, 2014

filtered nutrient solutions were added as $NaNO_3$, NH_4Cl and KH_2PO_4, each at a final concentration of $\sim 52\,\mu mol\,L^{-1}$.

Sterile filtered (0.2 µm, precleaned, Minisart 16534, Sartorius) solutions were added as substrates: (i) glucose (final concentration: $\sim 320\,\mu mol\,C\,L^{-1}$), (ii) algae-DOM derived from a culture of the haptophyte *Isochrysis galbana* (final concentration: $\sim 140\,\mu mol\,DOC\,L^{-1}$) and (iii) ^{13}C-labelled glucose (final concentration: $\sim 45\,\mu mol\,C\,L^{-1}$). The second addition of glucose (at day 699) was to a final concentration of $\sim 170\,\mu mol\,C\,L^{-1}$. *I. galbana* was grown in f/2 medium (Guillard and Ryther, 1962; Guillard, 1975; https://ncma.bigelow.org/node/79), to high density and cells were removed by sequential filtration (Sartobran 300: 0.45 µm followed by Minisart, Sartorius: 0.2 µm). Three liters of this filtrate were added to the respective treatments (Table 1).

All samples, except the two control samples ^{c}[Glu] and ^{c}[exud] were incubated with 1 L inoculum (3 µm filtrate, PC, Nuclepore), collected in the Weddell Sea (Antarctica) at a water depth of 100 m (12 December 2004; 67°49′ S, 55°33′ W; R/V *Polarstern*, PS67/006-118) and stored dark at 0 °C until the beginning of the experiment five months later. Inoculum used for the sterile controls ^{sc}[none], ^{sc}[Glu], and ^{sc}[exud] was sterile-filtered using Teflon filters (0.2 µm, Polycap, Whatman). All bottles were topped off to a total volume of 49 L with sterile filtered ultrapure water, and sealed with a rubber plug (Fig. 1). All bottles were dark-incubated at 0 °C in a cold room.

2.3 Sampling

During the first phase of the experiment (starting at 20 April 2005) each bottle was subsampled for bacteria, TEP, DOC, POC, glucose, and amino acids at eleven time points (days 2, 4, 6, 9, 13, 16, 21, 27, 34, 41, 55). Prior to sampling, each bottle was mixed for 2 min. to ensure sample homogenization. Sampling was performed using a glass tube which was installed through the cork of the bottle and sealed with a valve (Fig. 1). The water was sucked up using a peristaltic pump and Teflon tubing. The first ~ 100 mL of sample were discarded. Sterile air exchange in the headspace of each bottle was

BGD
11, 3065–3111, 2014

enabled by a filter (0.2 µm, Minisart 16534, Sartorius) which was also inserted into the cork.

After 70 days, the first phase of the experiment was terminated (Table 1). The remaining volume of all controls and one treatment of each type, i.e. [exud], [Glu], and [^{13}Glu], was filtered (0.2 µm, Polycap, Whatman) and solid-phase extracted (PPL, Varian). The four remaining bottles were incubated another 625 days (subsampling at 303/323 days). One [Glu] and the second [^{13}Glu] treatment as well as half of the remaining [Glu] and [exud] sample volumes were harvested at day 695 ending phase 2. The two remaining treatments [Glu] and [exud] received a second addition of glucose on day 699 (phase 3) and were incubated for another 35 days.

At every sampling day, unfiltered and filtered (0.2 µm; precleaned, Minisart 16534, Sartorius) samples (15 mL each) were collected for instantaneous organic carbon analyses. Three additional unfiltered samples (15 mL each) were filled into precombusted (500 °C, 5 h) glass ampoules (Wheaton) and frozen for later analysis of total organic carbon (TOC) and total dissolved nitrogen (TDN). Filtered samples for amino acid analyses (0.2 µm, 10 mL) were filled into precombusted ampoules, 10 M HCL (1 : 1, v : v, suprapur, Merck) was directly added and stored frozen (−28 °C) in the dark. 40 mL of samples were filtered (0.45 µm, GMF, Whatman), filled into PE bottles and stored frozen (−28 °C) for later nutrient measurements. Approximately 1 L of sample water was filled into PE bottles, filtered (0.4 µm PC; Poretics) and stained immediately for TEP analysis. For bacterial counts 60 mL of sample were filled into PE bottles and fixed with formaldehyde (10 % final concentration); replicate filters were prepared and counted within 1–4 weeks from these.

2.4 DOM extraction

Samples for DOM extraction were collected after 70 or 695 days (Table 1) by sequential filtration (1 µm, 0.2 µm, Whatman Polycap 75 TF) of the remaining sample in each bottle (between 16 L and 34 L), followed by acidification to pH 2–3 using HCl (Merck, Suprapur) and extraction (Dittmar et al., 2008) using pre-cleaned solid phase extrac-

BGD
11, 3065–3111, 2014

tion (SPE) cartridges (PPL, BondElut, 5 g). The extraction was performed by gravity at a speed of $< 12\,mL\,min^{-1}$. The cartridges were eluted with 20 mL or 40 mL MeOH (Merck, LiChrossolv), depending on sample volume, equivalent to an average enrichment factor of 750. Extracts were frozen (−28 °C) in precombusted (500 °C, 5 h) glass ampoules.

2.5 Bacterial and flagellate abundance

Bacteria were filtered onto 2 replicate black filters (0.2 µm Polycarbonate, Poretics), stained with 4′,6-diamidino-2-phenylindole (DAPI) or N′,N′-dimethyl-N-[4-[(E)-(3-methyl-1,3-benzothiazol-2-ylidene)methyl]-1-phenylquinolin-1-ium-2-yl]-N-propylpropane-1,3-diamine (SYBR Green), stored frozen and counted within 1–4 weeks using a Zeiss fluorescence microscope (for details see Porter and Feig, 1980; Noble and Fuhrman, 1998). At least 300 bacteria per replicate filter were counted in at least 10 to 20 fields of vision. Bacteria were enumerated in 2 size classes (< 2 µm and > 2µm) in samples from days 34 and 323.

The presence of flagellates was investigated on day 28 in 2 sub-samples ([exud] and [Glu]). Samples were filtered onto black filters (0.8 µm Polycarbonate, Poretics), stained immediately with DAPI, and counted via fluorescence microscopy (Kemp et al., 1993). Both test samples were negative.

2.6 Transparent exopolymer particles

Transparent exopolymer particles (TEP) were filtered directly after sampling (15 to $250\,mL\,filter^{-1}$) in quadruplicates onto 0.4 µm filters (PC, Poretics), stained with Alcian Blue and stored frozen until colorimetrical analysis 1–10 days later. The Alcian Blue staining capacity was calibrated with xanthan gum and quantified based on the absorptivity at 787 nm. Concentrations are given in µg xanthan equivalents per liter (Passow and Alldredge, 1995).

BGD
11, 3065–3111, 2014

2.7 Dissolved and particulate organic carbon and nitrogen

DOC, total dissolved nitrogen (TDN), and total organic carbon (TOC, unfiltered sample) were determined by high temperature catalytic oxidation and subsequent non-dispersive infrared spectroscopy and chemiluminescence detection (TOC-VCPN, Shimadzu). Final TOC, DOC and TDN concentrations are average values of triplicate measurements. If the standard variation or the coefficient of variation exceeded 0.1 µM or 1 %, respectively, up to 2 additional analyses were performed and outliers were eliminated. After each batch of five samples one reference standard (DOC-DSR, Hansell Research Lab, University of Miami, US), one ultrapure water blank and one potassium hydrogen phthalate standard were measured. The limit of detection (3σ of the blank) and quantitation (9σ of the blank) was 7 and 21 µmol C L^{-1}, respectively. The accuracy was ±5 %. POC was determined by the difference between TOC and DOC measurements.

2.8 Inorganic nutrients, free glucose, and total hydrolysable amino acids

Nutrients were measured using an Autoanalyzer (Evolution III, Alliance Instruments) according to seawater standard methods (Kattner and Becker, 1991; Grasshoff et al., 1999). Free glucose was analysed using high pressure anion exchange chromatography with pulsed amperometric detection based on previous methods (Johnson and LaCourse, 1990; Mopper et al., 1992; Engbrodt and Kattner, 2005). Samples were injected using an autosampler (AS-4000, Merck-Hitachi) and an ion chromatography system (DX-500; PA-1 guard column, 4 × 250 mm anion-exchange PA-1 column, ED-40 electrochemical detector, all Dionex). The detection limit was 20 nM C. Total hydrolysable amino acids were determined based on the method by Fitznar et al. (1999).

BGD
11, 3065–3111, 2014

2.9 Ultrahigh resolution mass spectrometry (FT-ICR-MS)

FT-ICR-MS analyses were carried out as described previously (e.g. Lechtenfeld et al., 2013). In summary, prior to analysis, DOM extracts were diluted with methanol: water (1 : 1, v/v). Samples were ionized by electrospray ionization (ESI, Apollo II electrospray ionization source, Bruker Daltonik, Bremen, Germany) in negative mode at an infusion flow rate of 120 µL h^{-1} on a Fourier transform ion cyclotron resonance mass spectrometer (FT-ICR-MS; SolariX, Bruker Daltonik, Bremen, Germany) equipped with a 12 T refrigerated actively shielded superconducting magnet (Bruker Biospin, Wissembourg, France). 300 scans were added to one mass spectrum. The magnitude threshold for the peak detection was set to a signal to noise ratio of ≥ 4. Mass spectra were recalibrated internally with compounds, which were repeatedly identified in marine DOM samples (Koch et al., 2008; Flerus et al., 2011; m/z: 247.06120, 297.13436, 327.14493, 369.15549, 397.15041, 439.16097, 483.18719, 551.24979, 595.23962). The average mass accuracy was below 100 ppb.

2.10 FT-ICR-MS data evaluation

All ions were singly charged as confirmed by the spacing of the related $^{12}C_n$ and $^{13}C^{12}C_{n-1}$ mass peaks. The spectra were evaluated in the mass range of 200–600 m/z. The base peak in this mass range was defined as 100 %, and relative intensities for all other peaks were calculated accordingly. For the process of formula assignment only peaks with a relative intensity between 2–100 % were considered. Molecular formulas were calculated from m/z values allowing for elemental combinations $^{12}C_{0-\infty}{}^{13}C_{0-1}{}^{1}H_{0-\infty}{}^{14}N_{0-4}{}^{16}O_{0-\infty}{}^{32}S_{0-2}{}^{34}S_{0-1}$ and a mass accuracy threshold of $|\Delta m| \leq 0.5$ ppm. The double bond equivalent (DBE = 1 + 1/2(2C − H + N)) of a valid neutral formula had to be an integer value ≥ 0 and the "nitrogen-rule" was applied (Koch et al., 2007). Formulas which were detected in a process blank (PPL extraction of ultrapure water) and in the list of potential surfactants (Lechtenfeld et al., 2013) were

BGD
11, 3065–3111, 2014

removed from the entire dataset. Formulas containing a ^{13}C or ^{34}S isotope and did not correspond to a parent formula (^{12}C, ^{32}S) were also removed from the dataset.

As an additional level of formula validation, all formulas were sorted according to DBE and ppm (Fig. 2a and b). A small proportion of formulas corresponded to very high DBE values, many of which were false assignments of sulfur containing compounds (as identified by the peak ratio of the parent and daughter ions). We therefore used DBE < 20 as an additional cut-off which resulted in an unambiguous assignment for the complete dataset. The distribution of mass accuracy also showed that the majority of the assigned formulas are well within the 0.5 ppm threshold. After these validation steps, we excluded the stable isotopes ^{13}C and ^{34}S because they only represented duplicates of the parent formulas for subsequent sample comparisons. Intensity weighted average (wa) molecular masses and element ratios were calculated from the normalized peak magnitudes. For formulas with a very high relative intensity, the isotope ratio provided an additional level of formula validation (Fig. 2c, Koch et al., 2007).

The degradation state was calculated using the degradation index (I_{DEG}), as suggested in a recent study (Flerus et al., 2012). I_{deg} can only be applied for PPL-extracted marine SPE-DOM analyzed with FT-ICR-MS and electrospray ionization in negative mode. I_{deg} can be calculated from raw peak magnitudes of ten compounds which were found to correlate either positively ($POS_{I_{deg}}$: $C_{13}H_{18}O_7$, $C_{14}H_{20}O_7$, $C_{15}H_{22}O_7$, $C_{15}H_{22}O_8$, $C_{16}H_{24}O_8$) or negatively ($NEG_{I_{deg}}$: $C_{17}H_{20}O_9$, $C_{19}H_{22}O_10$, $C_{20}H_{22}O_10$, $C_{20}H_{24}O_11$, $C_{21}H_{26}O_11$) with $\delta^{14}C$ (Flerus et al., 2012; Eq. 1):

$$I_{DEG} = \frac{\sum \text{magnitudes}\, NEG_{I_{DEG}}}{\sum (\text{magnitudes}\, NEG_{I_{DEG}} + \text{magnitudes}\, POS_{I_{DEG}})} \quad (1)$$

Higher I_{DEG} values correspond to a higher degree of degradation.

BGD
11, 3065–3111, 2014

2.11 Statistical analysis

In the following, duplicates will be presented as mean values and their respective range. Triplicates will be presented as means and the standard deviation of the mean. For the FT-ICR-MS dataset, we assessed the molecular similarity between samples by applying cluster analyses and multi-dimensional scaling (MDS) based on Bray Curtis similarity (Bray and Curtis, 1957) and untransformed normalized peak magnitudes (Software: "R" and Primer, Version 6).

3 Results

3.1 Controls

Four controls provided experimental validation (Table 1). The background control ^{c}[none] had an average DOC concentration of $15 \pm 3\,\mu mol\,DOC\,L^{-1}$ during the entire incubation period (Fig. 3). The DOC concentration of the sterile background control ^{sc}[none] was slightly higher ($23 \pm 12\,\mu mol\,DOC\,L^{-1}$). Both values were near the limit of quantitation. The DOC concentration of the inoculum was $43 \pm 1\,\mu mol\,L^{-1}$, but after the 1 : 49 dilution with the medium it contributed only marginally to DOC.

No temporal change in DOC (Fig. 3), nutrient, or TEP concentration was observed during the first 55 days in the two background controls ^{sc}[none] and ^{c}[none] or in the two sterile substrate controls, ^{sc}[Glu] and ^{sc}[exud]. The bacterial abundance increased slightly with time, but remained 2–3 orders of magnitude below the abundance of the substrate treatments (compare Fig. 5).

3.2 Inorganic nutrients

During the entire experiment, nutrient concentrations were sufficiently high to prevent limitation of bacterial growth (Fig. 4). Due to the addition of the algal-derived DOM solution, which contained nutrients, the initial concentrations of nitrate, nitrite and phos-

BGD
11, 3065–3111, 2014

phate were higher (~ 150 µmol L^{-1}, ~ 9 µmol L^{-1} and ~ 10 µmol L^{-1}, respectively) in the [exud] compared to the [Glu] and [^{13}Glu] treatments. The bacterial inoculum did not add detectable amounts of nutrients to the bottles.

Ammonium decreased from about 55 to 28 µmol L^{-1} in the [Glu] and [^{13}Glu] samples, slightly increased at 41 days and decreased again at 55 days whereas it remained almost constant at 60 µmol L^{-1} in the [exud] treatments. Nitrate, nitrite and phosphate remained almost constant in all treatments.

3.3 Substrate degradation and transformation

Glucose was completely metabolized after 21 days in [Glu] and [^{13}Glu] (Fig. 5a and c). Simultaneously, non-glucose DOC was generated with concentrations reaching 45 ± 3 ($[Glu]_{27}$) and 16 ± 2 µmol C L^{-1} ($[^{13}Glu]_{27}$; Table 2). This microbial produced DOC will be termed "non-labile" in the following. During the following 23 months, the non-labile DOC decreased at a rate of 11 nmol and 4 nmol C L^{-1} d^{-1} for $[Glu]_{27–695}$ and $[^{13}Glu]_{27–695}$, respectively (Table 2). The modification of DOM in the [exud] treatments could not be followed to the same detail, because the added exudate consisted already of a complex mixture of substances. The changes in DOC concentration in the [exud] treatments were small during the exponential growth of bacteria. After the exponential growth, the DOC in $[exud]_{27–695}$ decreased at a rate of 33 nmol C L^{-1} d^{-1} being faster than in the [Glu] treatments (Fig. 5b). Particulate organic carbon (POC) concentration in the [Glu] treatments reached its maximum at 21 days (30 ± 20 µmol C L^{-1}). Since the values reached the limits of precision (~ 5 % precision) the errors were relatively large. For the [exud] and [^{13}Glu] treatments POC was much lower. Except for the maximum concentration in $[exud]_{41}$ (12 ± 4 µmol C L^{-1}), POC could not be quantified in these treatments.

After 699 days glucose was added to one $[Glu]_{699}$ and one $[exud]_{699}$ treatment to track potential priming effects (Fig. 4). Twenty-five days later, the DOC concentration was slightly above the value before the glucose addition in both bottles and TEP had increased slightly to 100 and 109 µg X_{eq} L^{-1} in [Glu] and [exud], respectively.

BGD
11, 3065–3111, 2014

For selected samples total hydrolysable amino acids were determined. The proportion of organic carbon derived from amino acids (AA-C) increased for [Glu] and [^{13}Glu] and decreased in [exud] treatments at day 16 and increased towards the end of the first phase of the experiment (Table 3). The relative increase of AA-C with incubation time coincided with the average increase of organic nitrogen in the mass spectrometry data set (see Sect. 3.6).

3.4 Bacterial growth dynamics

Bacterial cell growth started slowly with a long lag phase (16 days) which may be attributed to the low incubation temperature (0 °C). Exponential growth and pronounced changes in substrate concentration and in TEP formation occurred between day 16 and day 21 in the [Glu] and [^{13}Glu] treatments and slightly later, between day 21 and day 26, in the [exud] treatments (Fig. 5). Despite the fact that the initial DOC concentration in [Glu] was 6.5 times higher than in [^{13}Glu], the maximum bacterial cell number was only slightly lower in [^{13}Glu]$_{34}$ compared to [Glu]$_{34}$ (Table 2). In contrast, the maximum bacterial cell number in [exud]$_{27}$ was 3.3 times lower than in [Glu]$_{34}$.

Not only were maximal bacterial cell concentrations in [exud] lower than in [Glu] but the cells were also slightly smaller. In [Glu]$_{34}$ 97 % of bacteria were > 2 µm, whereas only 92 % were that large in [exud]$_{34}$. After 323 days, 83 % and 63 % of bacteria were considered large in [Glu]$_{323}$ and [exud]$_{323}$, respectively. An estimation of the carbon content of a microbial cell was derived by dividing the POC concentration for [Glu]$_{21–41}$ and [exud]$_{27–41}$ (30 and 12 µmol POC L^{-1}, respectively) by the maximum bacterial abundance (2.8×10^{10} and 0.85×10^{10} cells L^{-1}, respectively). This resulted in an average cell carbon content of 13 fg C cell^{-1} for [Glu] and 17 fg C cell^{-1} for [exud].

The bacterial growth efficiency (BGE) can be calculated from the ratio of bacterial production (BP) and respiration (BR) using Eq. (2) (del Giorgio and Cole, 1998):

$$\mathrm{BGE} = \mathrm{BP}/(\mathrm{BP} + \mathrm{BR}) \quad (2)$$

BGD
11, 3065–3111, 2014

We estimated BGE (BGE_{estim}) by using the maximum POC concentration (POC_{max}) and the total amount of DOC consumed (DOC_{cons}; Eq. 3, Table 2):

$$BGE_{estim} = POC_{max}/(POC_{max} + DOC_{cons}) \quad (3)$$

BGE was comparable in the treatments which contained glucose (0.1) and substantially higher in the [exud] treatments (0.6).

Concentrations of flagellates were below detection in all treatments, suggesting that grazing did not significantly impact microbial dynamics.

3.5 Transparent exopolymer particles (TEP)

The concentration of TEP was highest during the exponential growth in $[Glu]_{21}$ and $[^{13}Glu]_{21}$ (1683 ± 189 and $1471 \pm 213\,\mu g\,X_{eq}\,L^{-1}$, respectively; Fig. 5). TEP production in [exud] treatments increased steadily till day 55, but remained an order of magnitude lower than in [Glu]. Although the bacterial abundance (BA) and TEP concentration were not correlated, the TEP/BA ratio at the maximum TEP concentration was similar for $[Glu]_{21}$ and $[^{13}Glu]_{21}$ (60 and 69 $fg\,X_{eq}\,cell^{-1}$, respectively). In contrast, this ratio was lower at 11 $fg\,X_{eq}\,cell^{-1}$ in the [exud] treatments. Similarly, the TEP/DOC ratio was appreciably higher in $[Glu]_{41}$ and $[^{13}Glu]_{41}$ (21 and 22 $\mu g\,X_{eq}\,\mu mol^{-1}\,C^{-1}$, respectively) compared to $[exud]_{41}$ ($0.8\,\mu g\,X_{eq}\,\mu mol\,C^{-1}$).

After the maximum TEP concentration was reached in the glucose incubations $[Glu]_{21}$ and $[^{13}Glu]_{21}$, TEP quickly decreased by about $25\,\mu g\,X_{eq}\,L^{-1}\,d^{-1}$ towards day 55. In the [exud] treatments, TEP increased until day 55 ($\sim 4\,\mu g\,X_{eq}\,L^{-1}\,d^{-1}$). At the end of the experiment, on day 695, TEP was below detection in all treatments.

3.6 Molecular formulas determined by ultrahigh resolution MS

The average molecular characteristics derived from ultrahigh resolution mass spectra of the solid-phase extracted treatments are presented in Table 4. The spectrum

BGD
11, 3065–3111, 2014

of $[^{13}Glu]_{70}$ was different from all other spectra: it generally showed fewer peaks but several additional peak clusters in the mass range above 600 m/z. The total peak magnitude was 2–3 times lower compared to all other samples. We therefore excluded this spectrum from the subsequent molecular comparisons.

To evaluate the data quality and reproducibility we compared ^{sc}[none], ^{c}[none], and ^{sc}[Glu], which only differed by the presence/absence of glucose. These controls were highly similar based on the number of assigned peaks, the I_{DEG}, and the peak magnitude weighted average element ratios (Table 4). The average coefficient of variation for their relative peak magnitude (as an indicator for reproducibility) was 7.9 %. The reproducibility for larger peaks (> 40 % relative peak magnitude) was better and resulted in a coefficient of variation of only 2.6 %. The average peak magnitude weighted ratios for the three replicates were $O/C_{wa} = 0.440 \pm 0.003$, $H/C_{wa} = 1.255 \pm 0.005$, $C/N_{wa} = 48.7 \pm 1.6$, and $C/S_{wa} = 234.8 \pm 7.5$. They were thus considered to be process replicates and their average peak magnitudes were used as a reference for the comparison with [Glu] treatments. ^{sc}[exud] was used as the control for the [exud] treatments.

The mass spectra of the different treatments revealed characteristic molecular differences particularly between the [Glu] and [exud] samples (Table 4). The [exud] spectra were characterized by an almost Gaussian peak distribution typical for natural organic matter (Fig. 6). In contrast, all [Glu] and $[^{13}Glu]$ treatments showed spectra with several additional peaks which did not match with the typical DOM peak magnitude distribution. The number of peaks and assigned molecular formulas in the [exud] spectra was higher compared to [Glu] treatments. The average molecule in the [exud] samples was larger and contained, compared to the number of C atoms, more oxygen and nitrogen. In addition, the [exud] treatments showed a lower I_{DEG} value (less degraded) than [Glu] treatments. I_{DEG} in the sterile control ^{sc}[exud] indicated that the original algae derived DOM was more labile than the background DOM introduced by the inoculum in ^{sc}[none], ^{c}[none] and ^{sc}[Glu].

BGD
11, 3065–3111, 2014

Incubation time also had an influence on the molecular composition. For [exud] treatments, the number of peaks increased with time. In [Glu] samples, the number of peaks increased at 70 days and decreased after 695 days. For all treatments, I_{DEG} was higher after 70 days and decreased again at 695 days. All treatments showed, compared to the number of C atoms, an increase in organic nitrogen and organic sulfur over incubation time. The total relative peak magnitude of the most stable compounds (island of stability, IOS, Lechtenfeld et al., 2014) compared to the total peak magnitude of all CHO compounds was calculated (Table 4). All treatments showed a lower relative contribution of IOS compounds after 70 days and a higher contribution towards 695 days.

The inoculum which was added to each treatment introduced a small proportion of refractory compounds (Antarctic surface water, 1 : 49 dilution), which needs to be considered for the molecular level comparison. Also, algae-derived DOM in the [exud] treatments contained a background of organic compounds derived from the culture medium. To avoid artifacts in data processing, molecular differences between treatments and controls were explored based on relative peak magnitude ratios: each mass peak in a substrate treatment was compared to the respective peak in the control treatment. Therefore, we used the relative peak magnitudes to calculate for each mass peak (= molecular formula), the peak magnitude ratio $Peak_{sample}/(Peak_{sample} + Peak_{control})$ resulting in a value reaching 1 for those peaks which were predominant in the sample treatment (red colors, Fig. 6) and a value near zero for peaks which were conspicuous in the control (blue colors, Fig. 6). In addition, we selected all peaks which were unique in the substrate treatments compared to their respective control (circles and numbers, Fig. 6). There were only few formulas (< 30) which uniquely occurred in the controls compared to the substrate incubations. The only exception was the ^{sc}[exud] control, in which we detected 173 unique formulas which were absent in the $[exud]_{70}$ treatment.

For data representation, we used the so-called van Krevelen diagram (van Krevelen, 1950; Kim et al., 2003; Fig. 6). All formulas which consisted of C, H, and O were displayed with respect to their molecular hydrogen/carbon and oxygen/carbon ratio.

BGD
11, 3065–3111, 2014

Saturated and reduced compounds appear in the upper left whereas unsaturated and oxidized substances plot in the lower right of the van Krevelen diagram.

Most molecular changes occurred in the boundary area of the patch in the diagram (indicated by the red color and circles in Fig. 6). For the [Glu] incubations, in particular, the strongest peak magnitude increase occurred outside of the center region in which the most persistent molecules would be displayed (as identified in Lechtenfeld et al., 2014, Fig. 6, black ellipse). However, after 695 days, these changes were less pronounced indicating that the material became more similar compared to the control samples. In the [exud] treatments, a relative loss of peak magnitude in the center of the patch was detected, particularly after 70 days (blue colors in Fig. 6) but relative peak magnitude gains also occurred outside of the center where non-refractory compounds would plot.

A hierarchical cluster analysis and multi-dimensional scaling (MDS) was applied to summarize the molecular differences between the treatments (Fig. 7). The analysis was based on untransformed relative peak magnitudes and did not include formulas containing ^{13}C. Although we expected that the ^{13}C-label in the [^{13}Glu] treatments would be detectable in the non-labile DOM pool, enrichment of ^{13}C compared to the unlabeled controls and treatments ^{c}[Glu], ^{sc}[Glu] and [Glu] was not found. This result was verified by an additional cluster analysis which included the ^{13}C-isotopes: the analysis yielded identical results as the approach in absence of the stable carbon isotopes (data not shown).

The degree of similarity between samples is indicated by the similarity scale in the cluster analyses (Fig. 7, left panel). A similarity value of 100 would be derived from two samples with identical relative intensities for all mass peaks in the spectrum. In the MDS analysis (Fig. 7, right panel), the similarity is expressed by the distance between samples in the MDS plot. A stress value of < 0.03 indicates an excellent representation of sample similarities in a two-dimensional representation. In addition, samples are grouped according to three levels of similarity derived from the cluster analysis.

BGD
11, 3065–3111, 2014

An additional confirmation of good data reproducibility of the measurements was provided by the high degree of similarity between the duplicates of the $[Glu]_{695}$ treatments (Fig. 7). In agreement with the results of the average parameters (Table 4), the three controls ^{sc}[none], ^{sc}[Glu] and ^{c}[none] showed the most similar molecular patterns (Fig. 7). These controls were most similar to the $[Glu]_{695}$ and $[^{13}Glu]_{695}$ treatments whereas $[Glu]_{70}$ was dissimilar. The [exud] treatments formed a cluster most dissimilar from all other samples and were similar to the respective control $^{sc}[exud]_{70}$.

4 Discussion

Marine DOC may be classified as "labile" if it is removed on time scales of hours to days, or as "semi-labile", if the lifetime reaches ~ 1.5 years and as "refractory" if it persists for 16 000 years and more (Hansell, 2013). In our study, the concentration of background DOC persisted until the end of the 2-year experiment, and could therefore be termed "semi-labile". However, using only bulk DOC concentration, we cannot decide whether the material produced in our experiment could persist on even longer time scales (refractory DOM). In fact, DOM can also be regarded as a dynamic continuum of compounds of variable persistence and degradability (Flerus et al., 2012; Lechtenfeld et al., 2014). Therefore the DOC that was microbially formed in the experiment will be called "non-labile" in this context.

DOM dynamics in [Glu] treatments could be followed in greater detail than in [exud] samples: in the [Glu] treatments the changes of the original substrate (glucose) could be directly monitored and, by that, labile and non-labile DOC could be distinguished. In the [exud] treatments we only observed the gross DOC change without the option to differentiate between substrate and non-labile DOC.

BGD
11, 3065–3111, 2014

4.1 Bulk DOM changes: microbial consumption and transformation of organic carbon

In both, [Glu] and [^{13}Glu] treatments microbial growth was most pronounced between day 16 and 27 and glucose was completely consumed by day 21, when DOC showed a local minimum. This data suggests that the labile glucose was utilized rapidly once bacteria responded, and non-labile DOM was generated, evident in our experiment as an increase of DOC in the absence of glucose. A similar shift from a mixture of bioavailable/non-labile DOM to non-labile DOM is assumed to have occurred between day 16 and 21 in the [exud] treatments, but it is less well resolved. The fraction of labile DOM was small in [exud] as DOC decreased by only 6 %. This suggests that the algae exudates which were added to the [exud] treatments were already degraded within the algae culture. Obviously the most bioavailable fraction of the exudation products was utilized immediately upon their release, and before the harvest of exudates for our experiment.

The rapid initial uptake of glucose and the production of non-labile DOC is consistent with findings of earlier studies (e.g. Brophy and Carlson, 1989; Ogawa et al., 2001; Gruber et al., 2006; Kawasaki and Benner, 2006) and the concept of the microbial carbon pump (Jiao et al., 2010; Benner and Herndl, 2011). About 10 % of the initial glucose carbon persisted as non-labile DOC in our experiment. This compares to a proportion of 5 % (incubation at 22° to 28 °C) found by Ogawa et al. (2001). Gruber et al. (2006) incubated much higher glucose concentrations resulting in a higher absolute non-labile DOC concentration, but a similar proportion of remaining DOC (4.4 %; incubation at 20 °C). The larger fraction of non-labile DOC in our experiment can be likely explained by the much lower incubation temperature of 0 °C. A positive correlation was previously found between the amount of the bioavailable DOM and temperature (Lonborg et al., 2010).

The dynamics of substrate consumption differed substantially between [Glu] and [exud] treatments: in [Glu] treatments, bacteria quickly metabolized 100 % of the glu-

BGD
11, 3065–3111, 2014

cose, at a decay rate of ~ 50 µmol glucose-C L^{-1} d^{-1} (day 16 to 21), producing large amounts of TEP and non-labile DOC in the process. A comparison between [^{13}Glu] and [Glu] suggests that the bacterial cell abundance and the amount of TEP produced by bacteria were independent of the available glucose concentration. In the [exud] treatments, the production of TEP and microbial biomass was smaller than in the [Glu] treatments and as much as 80 % of the initial DOC was still present even after 2 years of incubation, indicating that only 20 % of the algae exudate was bioavailable to the microbial population. The decay rate of this [exud] DOM was however faster compared to the decay of the non-labile DOM generated from glucose, suggesting a higher degree of bioavailability of this fraction of the algae exudate. A lower I_{DEG} value in [exud] than [Glu] before and after incubation supports a higher bioavailability of the non-labile DOM in [exud] compared to the non-labile DOM generated from glucose.

It is conspicuous that, although the amount of initial carbon consumed differed strongly between the three different treatments, the bacterial cell abundance in all three treatments was in the same order of magnitude. Since we can rule out additional carbon fixation by other autotrophic organisms, three explanations appear possible: (i) additional carbon was fixed by chemoautotrophic bacteria, (ii) the size and carbon content of the bacterial cells and (iii) the bacterial growth efficiency differed between treatments.

A substantial uptake of ammonium was observed in the [Glu] and [^{13}Glu] treatments but not in the controls. However, neither a parallel increase in nitrate, which would be indicative for chemoautotrophy, nor a decrease in ammonia in [exud] treatments was observed, where cell production was high compared to carbon utilization. Therefore chemoautotrophy is unlikely to have contributed to organic carbon production.

It is to be expected that the composition of the bacterial communities developed differently due to the different substrates. The microscopic observations indicated that 8 % of the bacterial cells in the $[exud]_{34}$ treatments were smaller in size compared to the [Glu] samples. The average carbon content of bacteria were similar in [exud] and [Glu] treatments (17 vs. 13 fg C $cell^{-1}$) and both were in the range of marine bacteria (e.g.

BGD
11, 3065–3111, 2014

12.4 ± 6.3 fg C cell^{-1}, Fukuda et al., 1998). The estimated BGE was much higher in the [exud] compared to the [Glu] treatments: BGE in the [exud] samples resembled the high values previously measured in situ in the Weddell Sea (Bjornsen and Kuparinen, 1991; del Giorgio and Cole, 1998) and in incubations using algal exudates (Wiebe and Smith, 1977; del Giorgio and Cole, 1998). BGE for the non-labile DOM generated in the glucose treatments was much lower and in agreement with low values for highly oxidized and nitrogen-poor substrates (Vallino et al., 1996; del Giorgio and Cole, 1998). For our experiment, the differences in BGE of the microbial population accurately reflected the mismatch between the carbon utilization and the associated bacterial growth.

We hypothesized that the bioavailability of the non-labile DOM observed after ~ 700 days could be increased by the addition of labile substrates (co-metabolism, priming effect; Horvath, 1972; Bianchi, 2011). However, the addition of glucose after 699 days in our experiment did not enhance degradation of the non-labile material. Instead, the added glucose was quickly consumed and the concentration of non-labile DOC slightly increased. Thus, for our setup, we can reject the hypothesis that addition of glucose enhances degradation of non-labile material. The slight increase of non-labile DOM after glucose addition rather supports the idea that the formation of non-labile DOM is dependent on the substrate concentration. Alternatively, the increase might also be explained by a degradation of non-labile DOC and a subsequent overcompensation of newly formed non-labile DOC.

TEP were formed during the experiment, particularly from bacteria growing on glucose and less from those growing on the algae-DOM. Bacteria are known to form TEP or their precursors (Stoderegger and Herndl, 1999; Sugimoto et al., 2007). Due to their affinity to other particles (stickiness), TEP are important for the biological pump (Alldredge et al., 1993; Passow, 1994; Logan et al., 1995). The formation of aggregates increases sinking velocities of particles, allowing a larger fraction of carbon to reach the deep ocean before degradation. Our experimental results suggest, however, that the role of TEP for the biological pump could be limited by its residence time. Comparison of the average degradation rate of 25 µg X_{eq} L^{-1} d^{-1} or ~ 1 µmol TEP-C d^{-1} in

BGD
11, 3065–3111, 2014

the [Glu] treatments to a typical POC concentration in the surface ocean of roughly 1–10 µmol POC L^{-1} (e.g. Stramski et al., 2008; Neogi et al., 2012) suggests very short residence times in the order of hours to days. However, TEP composition and, by extension, their bioavailability vary depending on their formation history.

4.2 Molecular imprints of substrates and incubation time

The central objective of this study was to explore the molecular composition of DOM produced by marine bacteria. The results obtained by FT-ICR-MS, glucose and amino acid analyses are in agreement with many previous studies which demonstrated that microbial degradation leads to molecular transformation of DOM (e.g. Tranvik, 1993; Ogawa et al., 2001; Kujawinski et al., 2004; Gruber et al., 2006; Rossel et al., 2013). Based on the molecular formulas and their respective peak magnitudes, all treatments differed from their substrate controls and were distinguishable with respect to substrate type and incubation time (Fig. 6). In a previous study, we hypothesized that "the chemical characteristics which lead to refractory properties of DOM are similar, largely independent from the source material and mediated by microbial or photodegradation" (Koch et al., 2005). Here, we specifically wanted to verify if the material which was produced by microbes and persisted for two years resembled refractory organic compounds. The results demonstrated that the molecular signatures, even after 2 years of incubation, strongly differed between substrates. Therefore our previous very general hypothesis must be rejected and refined: in fact, the molecular formulas which dominated in [Glu] treatments after 70 days were primarily compounds known for their short ocean residence times (Flerus et al., 2012; Lechtenfeld et al., 2014) and therefore do not belong to the pool of refractory compounds. However, the molecular signatures observed in the [Glu] treatments after 695 days were more similar to the controls (Fig. 7) suggesting the conversion of the labile compounds observed after 70 days into DOM characterized by elemental compositions which match with refractory compounds. This indicates at least two steps of DOM degradation: within days very labile glucose was utilized and new DOM was generated that lasted for at least 70 days. Continued micro-

BGD
11, 3065–3111, 2014

bial activity then transformed this DOM into the refractory DOM observed after 2 years. This implies that the DOM generated in previous, shorter-term bacterial glucose experiments might have not reached a molecular composition which resembles refractory DOM (Gruber et al., 2006). The [exud] incubation, in contrast, resulted in a molecular composition which strongly differed from the refractory patterns. The majority of compounds which were unique or increased relative to the control were not identified as being part of typical refractory compounds, even after two years of incubation (Fig. 7).

All control samples, except ^{sc}[exud], showed high I_{DEG} values indicating an advanced state of degradation. The inoculum was the exclusive source of natural organic matter in the background or ^{sc}[Glu] controls and was derived from Antarctic surface water. The DOM from surface water in the Southern Ocean has been shown to be old (Druffel and Bauer, 2000), resulting in high I_{DEG} values (Lechtenfeld et al., 2014). In comparison, the [exud] samples incubations had much lower I_{DEG} values (more labile) which increased with microbial degradation. Apart from these general patterns, however, I_{DEG} did not show a clear trend. One possible explanation for the [Glu] treatments is that most molecular changes occurred in the boundary area of the patch in the van Krevelen diagram whereas the I_{DEG} formulas are located more in the center (Fig. 6). An alternative way to assess the DOM degradation state was to compare the total relative peak magnitude of the most stable compounds (IOS, Lechtenfeld et al., 2014) to the total peak magnitude of all compounds. This comparison showed a more coherent trend in which both substrates had a smaller percentage of IOS compounds after 70 days (= more labile) and a higher contribution after 695 days (= more degraded).

Apart from variations in compounds containing carbon, hydrogen, and oxygen, changes in the contribution of heteroatoms other than oxygen were also found: the contribution of molecules containing nitrogen and sulfur atoms increased in all treatments (Table 4). For the [Glu] and [^{13}Glu] treatments this was expected: the DOM was directly derived from bacteria or reworked microbial biomass which is characterized by low C/N ratios (Fukuda et al., 1998). The organic substrate in the [exud] treatments was also derived from fresh biomass (algae) and relatively low C/N_{wa} were expected.

BGD
11, 3065–3111, 2014

However, the initial C/N_{wa} ratio in the $^{sc}[exud]_{70}$ was comparable to the $^{sc}[Glu]_{70}$ control, which was an additional indication that the exudates were already partly degraded before they were added to the samples. Therefore it was reasonable that C/N_{wa} (and C/S_{wa}) decreased similarly to the glucose incubations.

Although we found, based on a principal component analysis (PCA, data not shown), that nitrogen and sulfur containing molecules contributed only little to the sample heterogeneity, their occurrence could be good indicator for microbial alteration in future studies. The increase of the heteroatom contribution by microbial activity (as determined with FT-ICR-MS) was also observed in other experiments and environments (Schmidt et al., 2009; Liao et al., 2012; Rossel et al., 2013).

The mass spectrometric technique which was applied in this study only yields molecular formulas and no chemical structures. Therefore, it is not possible to unambiguously designate a specific compound as being refractory solely based on elemental composition and its similarity to deep ocean DOM. Definite conclusions can only be drawn if the elemental composition of a substance differs from refractory compounds (such as in the $[Glu]_{70}$ treatment). In theory, the chemically relevant constitutional isomers of a molecular formula with a molecular size of ~ 400 Da can be immense (Hertkorn et al., 2008). However, recent studies indicate that the structural diversity in refractory material is probably limited to specific substructures and functional groups (Hertkorn et al., 2006; Witt et al., 2009; Hertkorn et al., 2013).

Unexpectedly the ^{13}C-isotope label (D-Glucose-1-^{13}C) was not detectable in the DOM of the [^{13}Glu] treatments. There are several potential explanations: (i) the C_1-carbon was preferentially mineralized, (ii) the labeled compounds were not extracted or not ionized and detected, (iii) the labelled compounds were below the detection limit, or (iv) were preferentially embedded in the microbial biomass. A similar loss of the stable carbon isotope label was reported previously (Longnecker and Kujawinski, 2011).

BGD
11, 3065–3111, 2014

4.3 Implications for the marine organic carbon flux

The molecular view derived from FT-ICR-MS allowed a comparison of the organic fingerprints of DOM from the three treatments, controls and previous data on refractory marine DOM (Koch et al., 2005; Hertkorn et al., 2006). In agreement with several previous studies (Nebbioso and Piccolo, 2013 and references therein), this demonstrates the strong potential of FT-ICR-MS to unravel sources and transformation processes in organic matter.

The cooling of water masses in the polar oceans is the most important mechanism for the advection of DOC into the deep ocean ($\sim 86\,\mathrm{Tg\,C\,yr^{-1}}$; Hansell et al., 2009). The formation of refractory compounds is a prerequisite for an efficient sequestration of organic carbon during this process. Only chemically stable compounds can be exported into the deeper ocean and stored for millennia. Generally, the experimental results supported the concept of DOM as being a consortium of organic compounds with continuous age (Flerus et al., 2012) and vast differences in ocean residence times which can be substantially longer than the bulk age of DOC (Lechtenfeld et al., 2014). If we only focus on bulk changes, our non-labile DOC removal varied between 4 and $12\,\mu\mathrm{mol\,C\,L^{-1}\,yr^{-1}}$. Compared to the lowest in situ net DOC removal rates of 0.003–$0.15\,\mu\mathrm{mol\,C\,kg^{-1}\,yr^{-1}}$ in the bathypelagic (Hansell et al., 2009 and references therein), this was still 2–3 orders of magnitude faster. With respect to the formation of refractory DOM our study led to ambiguous results: DOM derived from algal exudates showed molecular patterns which did not match with refractory DOM whereas the molecular signatures in the incubated [Glu] samples closely resembled refractory material after two years. Compared to the average residence time of refractory DOM in the ocean, this time span is extremely short and would not comply with a steady state scenario in marine DOC models (Hansell et al., 2009). If the non-labile DOC produced in the experiment (10 % of the substrate DOC) would be identical to refractory substances in the ocean this would create an unreasonably high flux to the refractory DOC pool. It is therefore likely, and has been shown in previous studies, that other mechanisms than

BGD
11, 3065–3111, 2014

molecular composition contribute to the DOC preservation and degradation: a pulse of additional carbon could facilitate degradation by exceeding a chemoreceptive threshold for prokaryotes (Jannasch, 1995; Kattner et al., 2011). On the other hand, if the finding that a higher substrate concentration leads to a higher concentration of refractory DOM in the ocean is true, this would have a strong impact on marine carbon fluxes. The reservoir of DOC in the ocean would depend on primary production. A scenario of lower marine primary production in a more stratified future ocean would also result in a smaller pool of carbon fixed in the refractory marine DOC (positive feedback mechanism).

An incubation time of two years is, of course, much shorter than the average residence time of refractory DOC in the deep ocean. Therefore, non-labile DOC in the environment can be further degraded and transformed by prolonged incubation, changes in the microbial community (Carlson et al., 2004; McCarren et al., 2010), or photodegradation (Gonsior et al., 2009; Rossel et al., 2013).

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BGD
11, 3065–3111, 2014

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BGD
11, 3065–3111, 2014

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BGD
11, 3065–3111, 2014

Table 1. Experimental setup: three different substrates were incubated in 50 L glass bottles for 70, 695 or 734 days, respectively.

Bottle	Medium (45 L)	Inoculum (1 L)	Glucose (320 µM C)	Algal exudate (140 µM C)	^{13}C glucose (45 µM C)	DOM extraction after (days)	Treatment/control
^{sc}[none]	×	> 0.2 µm	–	–	–	70	Sterile background control
^{sc}[Glu]	×	> 0.2 µm	×	–	–	70	Sterile glucose control
^{sc}[exud]	×	> 0.2 µm	–	×	–	70	Sterile exudate control
^{c}[none]	×	> 3 µm	–	–	–	70	Background control
[Glu]	×	> 3 µm	×	–	–	70	[Glu]
[exud]	×	> 3 µm	–	×	–	70	[exud]
[^{13}Glu]	×	> 3 µm	–	–	×	70	[^{13}Glu]
[Glu]	×	> 3 µm	×	–	–	695	[Glu]
[Glu]	×	> 3 µm	×	–	–	695	[Glu]; co-metabolism
[exud]	×	> 3 µm	–	×	–	695	[exud]; co-metabolism
[^{13}Glu]	×	> 3 µm	–	–	×	695	[^{13}Glu]

BGD
11, 3065–3111, 2014

Table 2. DOC concentrations for selected days, DOC consumed (DOC_{cons}), minimum of non-labile DOC produced (calculated from the DOC concentration at day 27 minus day 21; DOC_{prod}), maximum POC concentration (POC_{max}), maximum abundance of bacterial cells (BA_{max}), cell carbon content (= POC_{max}/BA_{max}), decay rate of non-labile DOC (period from day 27 until day 695) and the estimated bacterial growth efficiency (BGE_{estim}). n.d.: not determined.

	[Glu]	[^{13}Glu]	[exud]
Average DOC concentrations ($\mu mol\,C\,L^{-1}$):			
Day 2: Initial substrate	326 ± 9	53 ± 2	144 ± 0
Day 21: Local minimum	28 ± 8	11 ± 1	135 ± 0
Day 27: Local maximum	45 ± 3	16 ± 2	141 ± 1
Day 695: Final concentration	34 ± 3	15	115
DOC_{cons} until day 21 ($\mu mol\,C\,L^{-1}$)	326[a]	53[a]	9
Minimum non-labile DOC_{prod} ($\mu mol\,C\,L^{-1}$)	17 ± 6	5 ± 1	6 ± 0
POC_{max} ($\mu mol\,C\,L^{-1}$)	30 ± 20	n.d.	12 ± 4
BA_{max} ($cells\,L^{-1}$)	2.8×10^{10}	2.4×10^{10}	8.5×10^{9}
Calculated cell carbon content ($fg\,C\,cell^{-1}$)	13	n.d.	17
Decay rate for non-labile DOC ($nmol\,C\,L^{-1}\,yr^{-1}$)	11	4	33
BGE_{estim} ($DOC_{cons}/(DOC_{cons} + POC_{max})$; unitless)	0.1	n.d.	0.6

[a] Glucose was completely consumed after 21 days.

BGD
11, 3065–3111, 2014

Table 3. Total hydrolysable dissolved amino acids and their contribution to organic carbon (AA-C) in µmol L^{-1} and percent of total DOC (%) in the [Glu], [^{13}Glu], and [exud] treatments on day 4, 16, and 41.

	[Glu] (n = 3)		[^{13}Glu] (n = 2)		[exud] (n = 2)	
t (d)	AA-C	%	AA-C	%	AA-C	%
4	0.7	0.0	1.9	0.5	0.7	0.3
16	6.1	0.4	4.1	4.3	0.2	3.7
41	4.3	2.3	3.4	3.1	0.7	5.6

BGD
11, 3065–3111, 2014

Table 4. Mean data of the molecular characterization via FT-ICR-MS: number of identified peaks in the spectrum (signal to noise ratio ≥ 4), number of formulas assigned (n), peak magnitude weighted averages (wa) of the molecular mass ($Mass_{wa}$), double bond equivalent (DBE_{wa}), oxygen to carbon ratio (O/C_{wa}), hydrogen to carbon ratio (H/C_{wa}), carbon to nitrogen ratio (C/N_{wa}), and carbon to sulfur ratio (C/S_{wa}). The degradation index (I_{DEG}) indicates the relative state of degradation of marine SPE-DOM using electrospray ionization in negative mode (Flerus et al., 2012). IOS (%; island of stability, Lechtenfeld et al., 2014) is the total relative peak magnitude of the most stable compounds compared to the total peak magnitude of all CHO compounds.

Sample	Peaks (n)	n	$Mass_{wa}$	DBE_{wa}	O/C_{wa}	H/C_{wa}	C/N_{wa}	C/S_{wa}	I_{DEG}	IOS (%)
$^{sc}[none]_{70}$	11 941	2183	415	8.7	0.437	1.260	50.5	239	0.81	50
$^{c}[none]_{70}$	13 618	2289	426	8.9	0.443	1.254	47.7	226	0.81	52
$^{sc}[Glu]_{70}$	12 480	2232	422	8.9	0.440	1.250	47.9	239	0.81	51
$[Glu]_{70}$	14 027	3404	449	8.9	0.433	1.296	39.2	175	0.85	43
$[Glu]_{695}$	13 591	2930	416	8.8	0.438	1.240	32.6	103	0.80	50
$[Glu]_{695}$	13 643	2565	420	8.7	0.439	1.260	37.9	118	0.82	50
$[^{13}Glu]_{695}$	13 357	2350	428	8.9	0.455	1.250	41.4	192	0.83	53
$^{sc}[exud]_{70}$	15 668	3082	412	8.2	0.454	1.291	45.7	182	0.52	40
$[exud]_{70}$	13 962	4424	494	9.9	0.462	1.269	33.8	153	0.70	36
$[exud]_{695}$	14 040	4677	442	9.0	0.459	1.261	35.5	155	0.61	39

BGD
11, 3065–3111, 2014

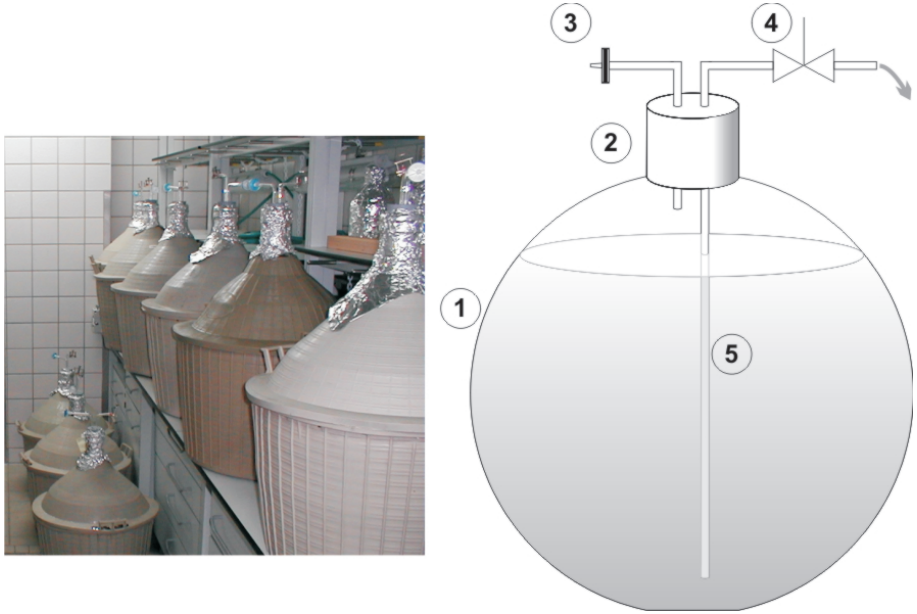

Fig. 1. Experimental setup: 11 glass bottles (1, total volume 49 L) each of which contained sterile artificial seawater medium (including additional nutrients), 1 L seawater inoculum and three different substrates. The bottles were closed with a rubber lid (2) and air exchange was enabled by a sterile 0.2 µm filter (3). Samples were taken through a valve (4) using a peristaltic pump and glass (5) and Teflon tubing.

BGD
11, 3065–3111, 2014

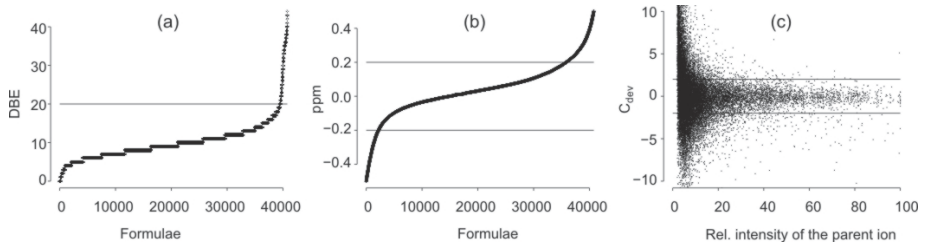

Fig. 2. Validation of molecular formulas: **(a)** double bond equivalent (DBE) and **(b)** mass accuracy (in ppm) are displayed in increasing order for molecular formulas assigned in the complete dataset. The quality of the peak magnitude ratio of a ^{12}C-parent formula vs. the corresponding ^{13}C daughter formula ($^{13}C_1\,^{12}C_{n-1}$) was calculated as **(c)** the difference between the number of carbon atoms in the assigned formula and the number of carbon atoms estimated from the stable carbon isotope ratio (C_{dev}, Koch et al., 2007). C_{dev} is displayed vs. the relative intensity of the parent ion.

BGD
11, 3065–3111, 2014

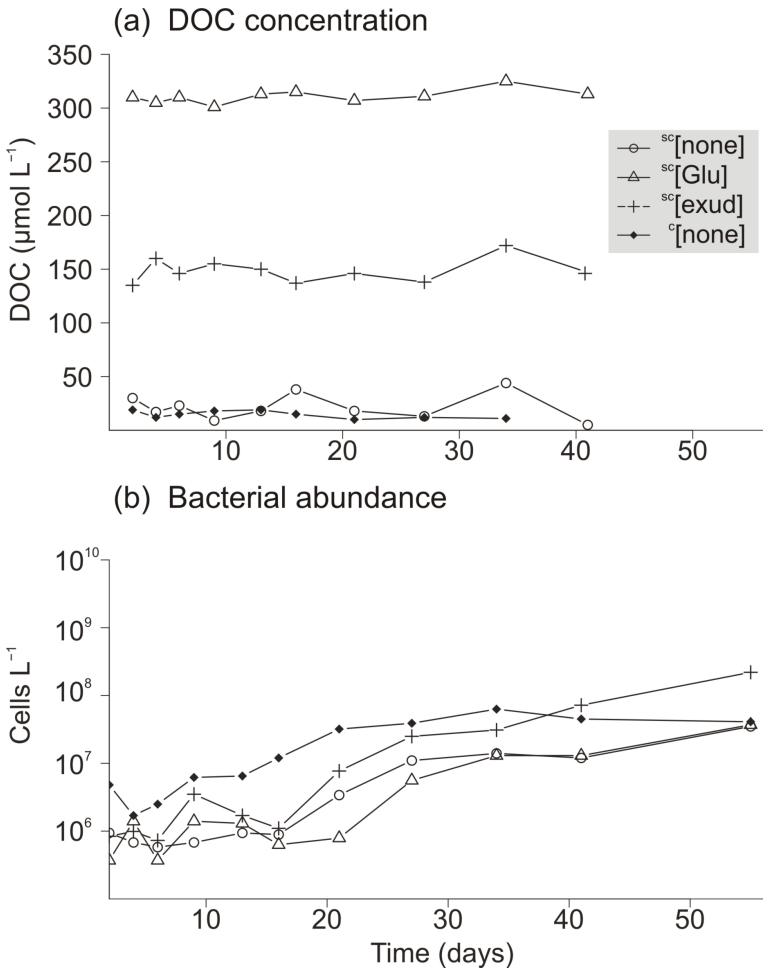

Fig. 3. Control samples: DOC concentration and bacterial cell abundance during the incubation period.

BGD
11, 3065–3111, 2014

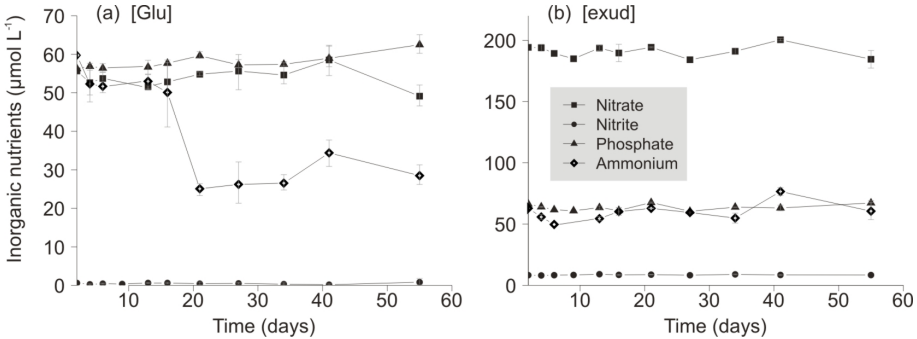

Fig. 4. Inorganic nutrients: concentration of nitrite, ammonium, phosphate and nitrate during the incubation period. Changes are displayed as averages for **(a)** [Glu] treatments ($n = 3$) and **(b)** [exud] treatments ($n = 2$). Error bars represent the range of values. Changes in [^{13}Glu] treatments (not shown) were almost identical to [Glu] samples.

BGD
11, 3065–3111, 2014

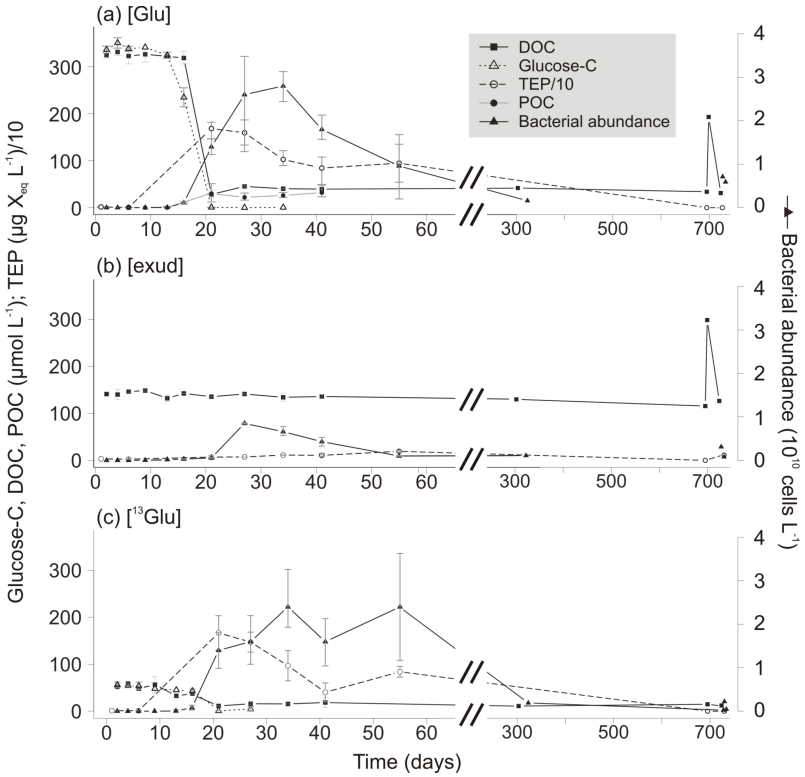

Fig. 5. Concentration changes with time for the microbial degradation of **(a)** glucose [Glu], **(b)** algal-derived exudates [exud] and **(c)** ^{13}C labeled glucose [^{13}Glu] treatments: dissolved organic carbon (DOC), particulate organic carbon (POC, only available for [Glu] treatments), glucose, transparent exopolymer particles (TEP), and bacterial abundance (bac, right y-axis). Error bars represent the range of values based on triplicates for [Glu] and duplicates for [exud] and [^{13}Glu]. On day 699, ~ 170 µM glucose-C was added to one [Glu] and one [exud]-bottle (singular samples).

BGD
11, 3065–3111, 2014

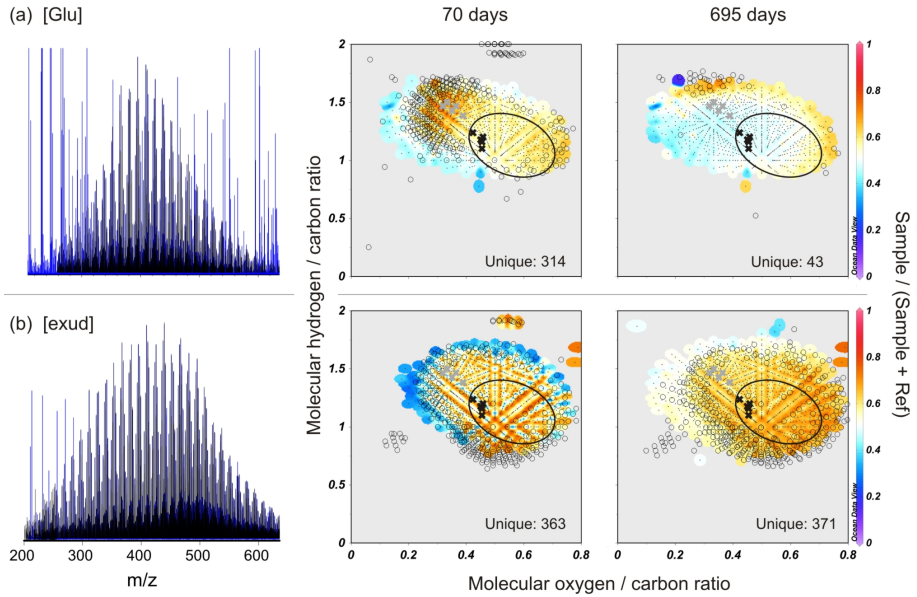

Fig. 6. FT-ICR mass spectra of the solid-phase extracts of microbially degraded **(a)** glucose [Glu] and **(b)** algal-exudate [exud] after 70 and 695 days of incubation. Left panel: measured spectra (in blue) and reconstructed spectra based on all identified molecular formulas containing C, H, and O (in black). Right panel: Van Krevelen plots represent all CHO molecular formulas. Formulas which occurred uniquely in the samples and not in the controls (average of (^{sc}[none], ^{sc}[Glu], ^{c}[none]) for [Glu] and ^{sc}[exud] for [exud]) are marked with a black circle. All other formulas are represented by color which reflects the ratio of the peak magnitude in the sample vs. the respective magnitude in the controls (Sample/(Sample + Ref)). The black circle represents the area in which molecular formulas with the highest residence times in the ocean would be displayed (according to Lechtenfeld et al., 2014; island of stability). The crosses represent those 10 peaks which are used to calculate the degradation state of marine DOM (Flerus et al., 2012; I_{DEG}): grey crosses represent molecular formulas which are labile; black crosses represent refractory molecular formulas.

BGD
11, 3065–3111, 2014

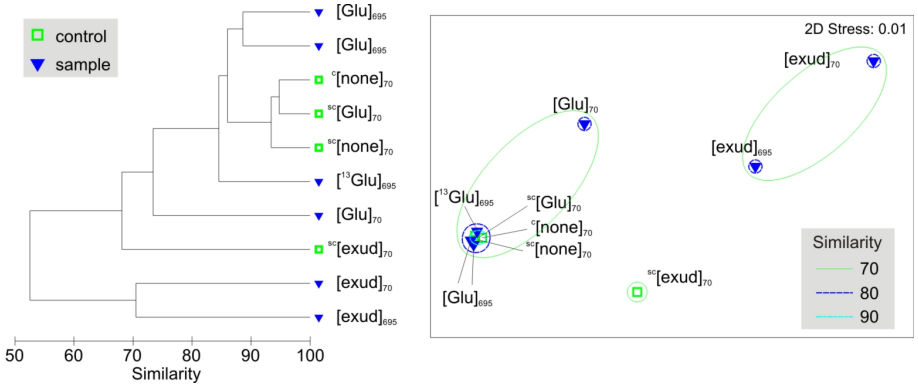

Fig. 7. Sample comparison based on hierarchical cluster analysis (left panel) and multi-dimensional scaling (right panel). Relative peak magnitudes of all identified molecular formulas (except stable isotopes) were compared based on Bray Curtis similarity and group average clustering.

BGD
11, 3065–3111, 2014