POTENTIAL IMPACTS OF CABLE BACTERIA ACTIVITY ON HARD-SHELLED BENTHIC FORAMINIFERA: A PRELUDE TO IMPLICATIONS FOR THEIR INTERPRETATION AS BIOINDICATORS OR PALEOPROXIES

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ABSTRACT

Cable bacteria (CB) are filamentous bacteria coupling sulphide oxidation to oxygen reduction over centimetre distances. This bacterial activity generates a strong pH gradient within the first few centimetres of the sediment that affects the microhabitats occupied by benthic foraminifera. Hard-shelled foraminifera are protists able to build a calcareous or agglutinated shell (called “test”). Here we study the impact of sediment acidification induced by CB activity (CBA) on calcareous test preservation. For this study, sediment cores were sampled in the macrotidal Auray estuary located on the French Atlantic coast. Living and dead foraminifera were quantified (until 5-cm depth) and discriminated using the Cell-Tracker™ Green vital marker. CBA was assessed with pH and oxygen profiles combined with quantitative Polymerase Chain Reaction (q-PCR). On two different intertidal mudflats, volumetric filament densities have been measured. They were comparable to those observed in the literature for coastal environments, with 7.4 ± 0.4 and 74.4 ± 5.0 m.cm⁻³ per bulk sediment respectively. Highly contrasting CBA (from low to very intense) were described with lowest pH at 5.8. This seems to lead to various dissolution stages of the foraminiferal calcareous test from intact to fully dissolved tests revealing the organic lining. The dissolution scale is based on observations of living Ammonia spp. and Haynesina germanica specimens under a Scanning Electronic Microscope. Furthermore, dead foraminiferal assemblages showed a strong calcareous test loss and an organic lining accumulation throughout depth under low pH, hampering the test preservation in deep sediment. These changes in both living and dead foraminiferal assemblages imply that CB must be strongly considered in ecological monitoring and historical studies using foraminifera as bioindicator and paleoenvironmental proxy.
1 INTRODUCTION

Cable bacteria (CB) were discovered by Pfeffer and co-workers in 2012. They are sulphide-oxidizing filamentous multicellular procaryotes from the Desulfobulbaceae family. They live in marine and freshwater sediments all around the world (Risgaard-Petersen et al., 2015; Burdorf et al., 2017). They inhabit a several centimetres thick zone from the oxic surface to the deep sulphidic sediment. CB generate a vertical bioelectrical current by coupling the cathodic oxygen or nitrate reduction at the sediment surface to the anodic sulphide oxidation at depth (Nielsen et al., 2010; Pfeffer et al., 2012; Risgaard-Petersen et al., 2012; Marzocchi et al., 2014). CB activity (CBA) strongly affects sediment geochemistry and results in a clear geochemical fingerprint: an oxygen decrease in the surface sediments combined with a pH maximum in this oxic zone, followed by a strong acidification of the pore water in the suboxic zone (Nielsen et al., 2010; Risgaard-Petersen et al., 2012, 2014; Meysman et al., 2015). It leads to iron sulphide and carbonate dissolution from the suboxic zone (Risgaard-Petersen et al., 2012; Rao et al., 2016; van de Velde et al., 2016) and possibly the calcareous shell of benthic organisms.

Benthic foraminifera are unicellular meiofaunal organisms. Most species can build a hard-shell (called a test) that can be agglutinated (cemented grains), hyaline calcareous (calcium carbonate) and porcelaneous calcareous (calcium carbonate enriched in magnesium). Benthic foraminifera are very abundant in marine areas (Martin, 2000) including transitional environments (Alve & Murray, 1999; Debenay et al., 2006). These systems located between marine and continental areas (i.e. littoral and estuarine zones), are subjected to a high variability of environmental factors (e.g. tide, freshwater flows, evaporation, development of seagrass meadows over seasonal cycles...). Then, benthic foraminifera are submitted to strong variability of physical and geochemical parameters such as temperature, salinity or pH that they must tolerate. Despite such variability, benthic foraminifera assemblages have been used in transitional environments as bioindicators for biomonitoring ecological state (Mojtabahid et al., 2006; Balsamo et al., 2012; O’Brien et al., 2021; Fouet et al., 2022) and as paleoenvironmental proxies to understand past ecosystems functioning (Martin, 2000; Murray, 2006; Katz et al., 2010; Keul et al., 2017; Durand et al., 2018). However, species with a calcareous test can be affected by low pH and carbonate undersaturation leading to test dissolution (Le Cadre et al., 2003; Bentov et al., 2009; de Nooijer et al., 2009; Haynert et al., 2011, 2014; Kurtarkar et al., 2011; Charrieau et al., 2018b). Even if they are rarely observed in situ, few studies have reported signs of severe test dissolution in living assemblages (e.g., Alve and Nagy, 1986; Buzas-Stephens, 2005; Polovodova and Schonfeld, 2008; Haynert et al., 2012; Cesbron et al., 2016; Charrieau et al., 2018a; Schönfeld and Mendes, 2022). These authors attribute these dissolution observations to low pH and undersaturation of the carbonate
system, which would be due to abiotic conditions (anthropogenic pollution, freshwater intrusions) or more rarely to biotic ones (degradation of plants). Under laboratory conditions, Le Cadre et al (2003) have shown that test dissolution of living *Ammonia becarri* starts at pH 7.5.

Benthic foraminifera live mainly in the topmost sediment. CB develop also on the few topmost centimetres of the sediment, which can therefore lead to an environmental overlap of the bacterial and foraminiferal communities. Richirt et al 2022 hypothesised that CBA induces the dissolution of calcareous tests within the sediment of the Lake Grevelingen (Netherlands). In the present study, we assess the impact of cable bacteria activity on the foraminiferal test preservation in sediment, testing the hypothesis that CBA is responsible for depleting the preservation of calcareous foraminifera in benthic assemblages. To achieve this, CBA was characterized by oxygen and pH microprofiling and CB density quantified by qPCR on intertidal mudflats of the Auray estuary (French Atlantic coast). Calcareous test dissolution stages were defined and quantified thanks to the analyse of SEM images. Then, we described living and dead foraminiferal assemblages to assess the calcareous test loss.
2 MATERIALS AND METHODS

2.1 Studied Area

The Gulf of Morbihan (Atlantic coast, France) is an enclosed marine bay where the Auray river flows. The Auray estuary is a macrotidal estuary with a tide range about 4 m (Figure 1).

Saltwater flows upstream over 20 km from the mouth of the estuary (357 m wide) which is tide-dominated (online data from OFB and IFREMER, accessed on May 05th 2022). The extensive description of this area was made by Fouet et al. (2022).

In September 2020, three stations along the Auray estuary were sampled on intertidal mudflats at low tide (Figure 1 and Table 1): station 1 (Plessis), station 2 (Fort Espagnol) and stations 3 (Kerouarc’h). Characteristics of the sampled stations are presented in Table 1.

<table>
<thead>
<tr>
<th>STATION</th>
<th>COORDINATES</th>
<th>DISTANCE FROM SEA</th>
<th>T (°C)</th>
<th>SALINITY</th>
<th>SEDIMENT DENSITY (g.cm⁻³)</th>
<th>VEGETATION COVER</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>47.646° N,</td>
<td>12 km</td>
<td>24.4</td>
<td>29.6</td>
<td>1.71 ± 0.12</td>
<td>Ulvea mat</td>
</tr>
</tbody>
</table>

Figure 1. Locations of sampling stations in the intertidal mudflats of the Auray estuary (France).
2.2 Sediment Sampling and Processing

One core was sampled from each station by hand with a Plexiglas® tube (82 mm inner diameter, 50 mm depth) and was transported within an hour in a cool box to the field laboratory. Then, the cores were submerged in ambient seawater for at least two hours to retrieve in situ conditions before microprofiling.

After microprofiling, each core was sliced using a core pusher and two trowels. Slice thickness was 2 mm for the first 20 mm depth, and 10 mm up to 50 mm depth. Each sediment slice was treated with Cell-Tracker™ Green (CTG 5 CMFDA: 5-chloromethylfluorescein diacetate; Molecular Probes, Invitrogen Detection Technologies) to mark living benthic foraminifera by fluorescence (Bernhard and Bowser, 1996; Bernhard et al., 2006). One mg of CTG was dissolved in 1 mL of dimethylsulfoxide (DMSO). This solution was then pipetted into the flask containing the sediment slice and its volume of ambient water to get a final solution of CTG about 1 µM (Bernhard et al., 2006; Pucci et al., 2009; Langlet et al., 2013, 2014; Cesbron et al., 2016). Each sample was then incubated in dark at room temperature overnight and then fixed with ethanol 99% (Choquel et al., 2021). Eventually, the samples were sieved with tap water over 315-, 150-, 125- and 63-µm mesh screens. Samples were conserved in 99% ethanol.

DNA was extracted from sub-samples of sediment slices at stations 1 and 2. 1-2 g every second slice down to 18-mm depth were sampled with a heat-sterilized spatula and transferred to 2 ml Eppendorf tubes, then frozen at -20°C degrees. Samples were sent in dry ice (CO_2(g) at -50°C) to the Microbiology Institute of Biology in Aarhus University (Denmark) for qPCR analysis to quantify cable bacteria biomass.

2.3 Microsensor Profiling

Two Unisense© profiling systems were used simultaneously. One consisted of two oxygen Clark-type microsensors with a 50 µm tip (Revsbech and Jørgensen, 1986; Revsbech, 1989), and the other of a pH sensor with a 500 µm tip diameter (PH500, Unisense). They were both mounted on a motorized micromanipulator linked to a computer, and connected to a MultiMeter S/N. The increment was 50 µm until 3 mm for oxygen. It was 100 µm around the seawater-sediment interface (SWI) for pH, and it was adapted in real time according to the evolution of the observed profile until 50 mm depth. For each core, eight descents were managed for O₂,
for a total of 16 profiles, while only one profiling was done for pH. To calibrate the O$_2$
microsensor, two points were measured, with the 100% of oxygen saturation in the bubbling
seawater column, and the 0% into the anoxic part of sediment. To calibrate the pH
microsensor, 3 NBS buffers were used (values 4.0, 7.0, 9.2).

2.4 Living Foraminiferal Analyses
Counts of hard-shell benthic foraminifera were performed in wet conditions (water) on the >125
µm fractions using an epifluorescence stereomicroscope (Olympus SZX12 with a light source
CoolLED pE-100, emission wavelength λ = 470 nm). All specimens showing clear green
fluorescence were picked and identified. Remaining specimens were considered as dead. In
doubtful cases, particularly with agglutinated species, specimens were crushed to inspect
whether fluorescence was due to the presence of protoplasm, to the autofluorescence of the
dissolution sediments grains composing the test, or the presence of bacteria or nematodes living inside
(Langlet et al., 2013; Cesbron et al., 2016). Total foraminiferal densities were expressed per
50 cm$^2$ of sediment and foraminiferal densities for sediment layers per 10 cm$^3$ volume.

For the taxonomy of hard-shell foraminifera species, reference publications on
estuarine foraminifera (Feyling-Hanssen et al., 1972; Hansen et al., 1976; Murray et al., 1979;
Scott et al, 1980; Hayward et al, 2004; Schweizer et al, 2011; Camacho et al, 2015; Richirt et
al, 2019; Fouet et al., 2022; Jorissen et al., 2023), and the World Register of Marine Species
were used. The distinction between the Ammonia phylotypes (Richirt et al, 2019) being difficult,
on particular on the dissolved tests, the results will be discussed at the genus level.

2.5 SEM Imaging
Living foraminifera from three layers (0-2 / 6-8 / 40-50 mm depth), according to main pH
features, were all observed under a Scanning Electronic Microscope (SEM). Two different
high-resolution SEM were used: a DEBEN Hitachi TM4000 at the LPG (samples not
metallised, 15kV, wd = 6,5 mm, partial vacuum (60 Pa)) and a Zeiss EVO LS10 at the Service
Commun d’Imageries et d’Analyses Microscopiques of Angers University (SCIAM; samples
not metallised, 20 kV, wd = 6,5 mm, partial vacuum (60 Pa), amperage 200 to 250 pA). Few
scales of calcareous test dissolution of living foraminifera have been proposed in the literature
(Corliss and Honjo, 1981; Le Cadre, 2003b; Haynert et al., 2011; Gonzales et al., 2017;
Charrieau et al., 2018c; Schönfeld and Mendes, 2022). These authors proposed scales varying
from 4 to 5 different stages based on SEM images or stereomicroscope observations. They
used a wide variety of morphological criteria to describe each dissolution stage (i.e. the number
of calcite layers altered and chambers damaged, the presence of cracks or holes, whether the
inner organic lining was visible, etc.). In the present study, we propose a scale of six dissolution
stages based on SEM pictures of the two most abundant calcareous species in our living assemblages (*Ammonia* spp. and *Haynesina germanica*).

Table 2. Description of the six dissolution stages of the calcareous tests of *Ammonia* spp. and *Haynesina germanica*.

<table>
<thead>
<tr>
<th>Dissolution Stage</th>
<th>Name</th>
<th>SEM Observations and Stage Descriptions</th>
<th>Figures</th>
</tr>
</thead>
<tbody>
<tr>
<td>DS-0</td>
<td>Intact test</td>
<td>intact, glassy test with a smooth surface and cylindrical pores, no sign of dissolution.</td>
<td>Fig. 3-1</td>
</tr>
<tr>
<td></td>
<td></td>
<td>transparent test with cylindrical pores, alteration of the last calcite layer only, appearance of the interpore sutures in <em>H. germanica</em> (scarce in <em>Ammonia</em> spp., alteration more visible on the inter-chamber walls).</td>
<td>Fig. 4-1</td>
</tr>
<tr>
<td>DS-1</td>
<td>Slight surface dissolved test</td>
<td>dull, whitish test with some fusion of adjacent widen pores, calcite layers cracking and crumbling, last chamber often lost, thinner and blunt tubercular ornamentation of <em>H. germanica</em>.</td>
<td>Fig. 3-2</td>
</tr>
<tr>
<td></td>
<td></td>
<td>opaque and cracked test with a strong alteration of all calcite layers, brittle test with holes, fusion of widen pores, the organic lining can be visible, loos of last chamber, broken ornamentation of <em>H. germanica</em>.</td>
<td>Fig. 4-3</td>
</tr>
<tr>
<td>DS-2</td>
<td>Peeled test</td>
<td>nearly completely dissolved test, only the inter-chamber walls remaining, the last chambers often absent, dissolved peripheral chambers with the inner organic lining visible.</td>
<td>Fig. 3-4</td>
</tr>
<tr>
<td></td>
<td></td>
<td>totally dissolved test revealing the inner organic lining, may keep the foraminifera shape allowing the identification of the genus <em>Ammonia</em> (not observed for <em>H. germanica</em>).</td>
<td>Fig. 4-5</td>
</tr>
<tr>
<td>DS-3</td>
<td>Cracked test</td>
<td>nearly completely dissolved test, only the inter-chamber walls remaining, the last chambers often absent, dissolved peripheral chambers with the inner organic lining visible.</td>
<td>Fig. 3-5</td>
</tr>
<tr>
<td></td>
<td></td>
<td>totally dissolved test revealing the inner organic lining, may keep the foraminifera shape allowing the identification of the genus <em>Ammonia</em> (not observed for <em>H. germanica</em>).</td>
<td>Fig. 3-6</td>
</tr>
</tbody>
</table>
Figure 2. Dissolution scale of Ammonia spp. based on high-resolution SEM images (spiral view). The specimens are classified into six stages of test dissolution from intact (stage 0) to fully dissolved (stage 5). For stages 0 to 2, a zoom on the last formed chamber was done (1-b, 2-b, 3-b), and on the n-1 chamber for stage 3 (4-b). White arrows point the organic lining.
Figure 3. Dissolution scale of *Haynesina germanica* based on high-resolution SEM images. The specimens are classified into five stages of test dissolution from intact (stage 0) to the «star shape» (stage 4). No organic lining (stage 5) has been identified as belonging to the taxa *Haynesina*. For stages 0 and 1, a zoom on the last formed chamber was done (1-b, 2-b), and on the n-1 chamber for stages 2 and 3 (3-b, 4-b). White arrow points the organic lining.
2.6 Dead Foraminiferal Analyses

Non fluorescent tests of foraminifera were counted as dead specimens and picked in wet conditions (water) to preserve the organic linings from fully dissolved tests. We proceeded under a stereomicroscope (ZEISS Stemi sv11) in three sediment layers: the surface layer (0-2 mm), the subsurface layer (6-8 mm) and the deep layer (40-50 mm). After quick observations, when high densities were estimated (above 500 individuals; Patterson and Fishbein, 1989)) fractions were splitted into 8 sub-samples using a wet splitter (Charrieau et al., 2018a).

2.7 Ratios in Foraminiferal Assemblages

In order to characterize the loss of calcareous in the assemblages, we defined a ratio as follows:

\[ \frac{C}{T} = \frac{\text{calcareous foraminifera}}{\text{total foraminifera}} \]

Calcareous foraminifera are counted regardless their dissolution stage and total foraminifera include agglutinated individuals. To estimate the intensity of dissolution in the assemblage, we calculated the following ratio:

\[ \frac{DS-5}{C} = \frac{\text{calcareous test at dissolution stage 5}}{\text{total calcareous foraminifera}} \]

These ratios were calculated on both living and dead assemblages for layers 0-2 / 6-8 and 40-50 mm.

2.8 Statistical Procedure

The putative relationship between CBA and the advanced dissolution stages of the living calcareous test foraminifera was assessed by applying the parametric Fisher’s test followed by the pair-wise Fisher’s test for post-hoc comparisons were used. To minimize the risk type 1 error p-values were FDR-adjusted. The significance level was set to 5 %. As the last layer of calcite produced during the growth of the foraminifera covers the entire test and is thinner than the others (Haynes, 1981; Hansen, 1999; Debenay et al., 2000; Boudagher-Fadel, 2018), DS-1 and 2 are more commonly observed resulting from a process of gradual dissolution or precipitation of calcite. Discrimination of the effect of the dissolution process is therefore made on the alteration of several calcite layers as for DS-3 and above. For this purpose, the dissolution stages were combined into two groups: no to slight dissolution (DS-0, 1 and 2) and moderate to severe dissolution (DS-3, 4 and 5). These two groups were then compared between each of the three stations, and between the different depth levels (0-2 / 6-8 / 40-50 mm depth) for each station. Statistics were carried out using the $R$ software using the "stats" and "rstatix" packages.
2.9 Sediment Treatment for DNA Extraction and Quantification

DNA was extracted from weighed amounts of sediment (0.22 - 0.25 g wet weight). DNA extraction was carried out using DNeasy PowerLyzer PowerSoil Kit (Qiagen) and the DNA was collected in 60 μl elution buffer. The analysis followed the procedures outlined in Geelhoed et al. (2020). The primer combination of ELF645wF and CB836wR was used to target the 16S rRNA gene of the marine cable bacteria of the genus Candidatus Electrothrix Trojan, 2016. The calibration curves were obtained using a synthetic standard (sequence accession KR912339.1, position 611-912, synthesized by Eurofins Genomics, Denmark) diluted in a 10-fold dilution series. The standards and samples were run in triplicates. Each reaction contained the master mix (RealQ Plus 2x Master Mix Green, Low ROXTM, Ampliqon, Denmark), forward and reverse primers (0.2 μM), BSA (1 μM). The qPCR was performed with a real time PCR analyser (AriaMX, Agilent). The thermal cycles were as follows: 15 min at 95 °C for initial denaturation followed by 40 cycles of 15 s at 95 °C (denaturation), 30 s at 60 °C (annealing), and 20 s at 72 °C (amplification). Afterwards, the melting curve was obtained by 30 s at 95°C, 30 s at 60 °C, and 30 s at 95 °C. Finally, the temperature was held for 5 min at 40 °C to terminate the analysis. The results are reported as the unit gene copies (g wet sediment)⁻¹.

CB filament density were calculated as in Geelhoed et al. (2020), using data of wet sediment density from a previous campaign in 2019 (Table 1), and expressed in m.cm⁻³.

3 RESULTS

3.1 Microsensor Profiles and Cable Bacteria Abundance

Oxygen penetration depth in the sediment at stations 1, 2 and 3 was 1.4 ± 0.2, 0.9 ± 0.3 and 0.9 ± 0.2 mm, respectively. At station 1, pH rapidly decreased from 7.7 at the Sea Water Interface (SWI) to a minimum of 6.8 at 15 mm depth. Below this minimum, pH stabilised to 7.2 around 40 mm depth. In contrast, at stations 2 and 3, pH increased immediately below the SWI, from 7.8 to 8.1 at 0.8 mm depth and to 7.95 at 0.6 mm, respectively (Figure 4). Below these maxima, at both stations, pH reached a minimum of 5.8 at 7 mm depth at station 2 and of 6.3 between 7-19 mm depth at station 3. Below these minima, pH stabilised at 6.8 after 25 mm depth at station 2, and at 6.9 after 34 mm depth at station 3. Those profiles with an oxygen decrease in the surface sediments combined with a pH maximum in this oxic zone, followed by a strong acidification of the pore water in the suboxic zone, are typical CBA fingerprints.

At station 1, the number of 16S CB copies of Candidatus Electrothrix ranged from 0.23 ± 0.01 x10⁷ to 0.48 ± 0.01 x10⁷ 16S copies.(g wet sediment)⁻¹, and remained constant through depth (Figure 4). At station 2, it amounted to 2.8 ± 0.12 x10⁷ 16S copies.(g wet sediment)⁻¹ in the upper 5 mm of sediment and progressively decreased to about 0.3 ± 0.01 x10⁷ 16S
copies.(g wet sediment)$^{-1}$ in the 16-18 mm depth layer. The maximum 16S CB copies of Ca. Electrothrix in station 2 corresponded to the maximum pH in depth. According to Gelhoed et al. (2020) and using sediment density from the same stations obtained in 2019 (pers. comm. M. Fouet), we calculated a CB density of $7.4 \pm 0.4$ and $74.4 \pm 5.0$ m.cm$^{-3}$ at stations 1 and 2 respectively.

![Graph showing sediment oxygen (blue circles) and pH (orange diamonds) microprofiles at the three stations, and vertical distribution of cable bacteria abundance (qPCR of Ca. Electrothrix 16S rRNA gene copies, grey bars) for stations 1 and 2. 0 is the position of the Sea Water Interface (SWI). The vertical red line represents neutral pH. The oxygen profile presented is one of those obtained by microprofiling, and representative of O$_2$ penetration for each station.]

**Figure 4.** Sediment oxygen (blue circles) and pH (orange diamonds) microprofiles at the three stations, and vertical distribution of cable bacteria abundance (qPCR of Ca. Electrothrix 16S rRNA gene copies, grey bars) for stations 1 and 2. 0 is the position of the Sea Water Interface (SWI). The vertical red line represents neutral pH. The oxygen profile presented is one of those obtained by microprofiling, and representative of O$_2$ penetration for each station.

### 3.2 Hard-Shelled Benthic Foraminiferal

#### 3.2.1 Living Foraminiferal Diversities and Densities

The foraminiferal species assemblages were typical of the estuarine environments (Debenay et al., 2000), with a poor species richness (14, 13 and 18 species at stations 1, 2 and 3 respectively). *Ammonia* spp. and *Haynesina germanica* (Ehrenberg, 1840) both strongly dominated the assemblages at all three stations (25.1 and 51.5 % respectively of the total assemblage for station 1, 14.5 and 48.2 % for station 2, 7.3 and 61.4% for station 3; **Figure 5**). *Ammonia* spp. included the species *Ammonia veneta* (Schultze, 1854) (phytotype T1 after Hayward et al., 2004), *Ammonia aberdoveyensis* Haynes, 1973 (phytotype T2 after Hayward et al., 2004), and *Ammonia confertitesta* Zheng, 1978 (phytotype T6 after Hayward et al., 2004). Agglutinated foraminifera represent 19.9, 25.7 and 12.7 % of the total assemblage at
stations 1, 2 and 3, respectively. They were dominated by *Ammobaculites agglutinans* (d’Orbigny, 1846).

**3.2.2 Living Foraminiferal Vertical Distribution**

Total densities of CTG-labelled foraminifera in cores 1, 2 and 3 were 1273, 548, and 1431 ind.50cm$^{-2}$ respectively. Highest densities were found in the first layer of sediment (0-2 mm depth) for all cores with 295, 137 and 371 ind.10cm$^{-3}$ at stations 1, 2 and 3, respectively (**Figure 2**), where dioxygen was available and pH was maximal (**Figure 4**).

At station 1, total density dropped below 2 mm to stabilize at 30 ± 9 ind.10cm$^{-3}$ (**Figure 5**). At station 2, the vertical distribution of total densities showed two maxima. The first at the SWI and a second at 18-20 mm depth with 47 ind.10cm$^{-3}$. A first minimum of 11 ind.10cm$^{-3}$...
was observed at 8-10 mm depth close to the lowest pH layer and a second minimum of 5 ind.10 cm$^{-3}$ was observed at the bottom of the core. At station 3, after a maximum at the SWI, foraminifera density decreased gradually with depth, following the pH trend, to reach on average 19 ± 4 ind.10 cm$^{-3}$ from 20 to 50 mm depth.

At station 1, the ratio of calcareous foraminifera in the living foraminiferal assemblage (C/T) was 0.91 for the SWI (Table 3) and around 0.77 ± 0.07 for the layers below. At station 2, C/T was 0.97 of the SWI and on average 0.64 ± 0.16 between 2- and 50-mm depth (Appendix). However, agglutinated taxa dominated the assemblages from 10 to 18 mm, just below the pH minimum, with a drop of C/T ratio to 0.39 ± 0.18 (Appendix). At station 3, the C/T ratio was 0.97 at the SWI and decreased asymptotically as calcareous foraminiferal densities vanished to reach 0.72 ± 0.15 below 20 mm after the pH minimum zone (Appendix).

### 3.2.3 Calcareous Test Dissolution of Living Foraminifera

Figure 6 shows the dissolution stage (DS) of calcareous foraminifera for three selected layers (0-2 / 6-8 / 40-50 mm) for living assemblages. At station 1, living specimens with calcareous test showed low alteration. The DS remained stable through depth (p > 0.05). Specimens with “Intact tests” (DS-0) or “slight surface dissolved tests” (DS-1) represented 90% of calcareous foraminifera. The strongest dissolution stages were DS-2 (“peeled test”) and DS-3 (“cracked test”) accounting for less than 10%.

<table>
<thead>
<tr>
<th>St 1</th>
<th>Relative abundance (%)</th>
<th>St 2</th>
<th>Relative abundance (%)</th>
<th>St 3</th>
<th>Relative abundance (%)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>0</td>
<td>20</td>
<td>40</td>
<td>60</td>
<td>80</td>
</tr>
<tr>
<td>surface (0-2 mm)</td>
<td>0</td>
<td>20</td>
<td>40</td>
<td>60</td>
<td>80</td>
</tr>
<tr>
<td>subsurface (6-8 mm)</td>
<td>0</td>
<td>20</td>
<td>40</td>
<td>60</td>
<td>80</td>
</tr>
<tr>
<td>deep (40-50 mm)</td>
<td>0</td>
<td>20</td>
<td>40</td>
<td>60</td>
<td>80</td>
</tr>
</tbody>
</table>

Figure 6. Relative abundance of living benthic foraminifera with calcareous test for each dissolution stage for 10 cm$^{3}$ of sediment (Ammonia spp. and H. germanica from the >125 µm fraction). Three depth levels were analysed: the surface (0-2 mm; oxic zone), the subsurface (6-8 mm; suboxic zone corresponding to pH minimum), and the deeper (40-50 mm; anoxic zone). The numbers on the lower right of the boxes are the total numbers of SEM photographed specimens.

Conversely, at station 2, many foraminifera were very fragile under manipulation. Numerous “fully dissolved tests” (DS 5) with only the organic lining were observed through depth (50 ind.50 cm$^{-2}$; Appendix). At the SWI (0-2 mm layer), DS-0 and DS-1 tests represented 95% of the calcareous test foraminifera in the living assemblage. Only few DS-2 and DS-5 specimens were present. In the subsurface level (6-8-mm depth), corresponding to the most
acidic conditions, no DS-0 and DS-1 specimen were observed. DS-4 and DS-5 tests were about 40% of the calcareous tests observed. At the deepest layer (40-50 mm), DS-5 specimens were dominant (>50%). The surface layer was significantly different (p < 0.005) from the two deeper layers that showed no significant differences (p = 0.267).

At station 3, many foraminifera were fragile under manipulation, and DS-5 specimens were abundant through depth with about 140 ind.50cm$^{-2}$ (Appendix). At the SWI (0-2 mm layer) and in the subsurface level (6-8-mm depth), DS-0 and DS-1 specimens represented about 75% while DS-2 accounted for 20%. Few specimens of DS-3, DS-4 and DS-5 were observed. At the deepest layer (40-50 mm), DS-5 specimens were the most abundant calcareous tests foraminifera (78%). The severe DS (DS-3, 4 and 5) were significantly overrepresented in the deep layer than in the surface and subsurface layers (p < 0.005). DS were not significantly different between surface and subsurface (p = 1).

Overall, the exact Fisher’s test revealed significant difference dissolution stages among stations (p<0.005). The pair-wise Fisher’s exact test showed that the low DS (0,1,2) were significantly overrepresented at station 1 compared to the other two stations (p<0.005). Furthermore, there were no significant difference between stations 2 and 3 (p=0.532).

### 3.2.4 Calcareous vs. Agglutinated Foraminifera in the Dead Assemblages

Species in the benthic foraminiferal thanatocoenosis were the same as in the living assemblages. At station 1, calcareous taxa dominated agglutinated ones in the dead assemblage with C/T ratio varying from 0.74 to 0.89 (Table 3). The proportion of organic lining (DS-5/C) increased slightly with depth, from 0.06 to 0.18. On the other hand, at station 2, agglutinated taxa dominated the dead assemblage in the surface and subsurface levels (C/T ratio of 0.43 and 0.36 respectively) but not in the deepest one even if they remained abundant (0.65; Table 3). The DS-5/C ratio was very high in all three depth layers, remaining >0.70. At station 3, C/T ratio remained high in the dead assemblage of both surface and subsurface with 0.88, 0.83, and decreased strongly to 0.36 in depth where agglutinated specimens were dominant. The DS-5/C ratio increased with depth, from 0.06 at the surface to 0.95 in the deeper layer.

Comparing dead and living assemblages, it can be noted that for station 1, C/T ratio were not very different whatever the depth (Table 3). Stations 2 and 3 showed much lower C/T ratios in the dead assemblages indicating a marked loss of calcareous foraminifera during taphonomic processes although this difference is not significant. In addition, stations 2 and 3 showed a higher occurrence of DS-5 tests in the dead assemblages resulting in high DS-5/C ratios. In detail, station 2 showed the highest DS-5/C ratio in the subsurface layer (0.96) where pH is minimal, while station 3 showed a strong increase of this ratio in the deepest layer (0.95).
Table 3. Densities of living and dead foraminifera for each depth layer at the three all stations (ind.10 cm\(^{-3}\) of sediment). Depth correspondences: surface (0-2 mm), subsurface (6-8 mm) and deep (4-5 mm). "Calcareous" class includes the DS-5 specimens (fully dissolved test showing the organic lining). A = agglutinated, C = calcareous, ratios are those described in the methods.

<table>
<thead>
<tr>
<th>Living foraminifera</th>
<th>Dead foraminifera</th>
</tr>
</thead>
<tbody>
<tr>
<td>Agglutinated (A)</td>
<td>C/T ratio</td>
</tr>
<tr>
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</tr>
<tr>
<td>subsurface [6-8 mm]</td>
<td>3 36</td>
</tr>
<tr>
<td>deep [40-50 mm]</td>
<td>6 26</td>
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<td>surface [0-2 mm]</td>
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<td>7 137</td>
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<tr>
<td>deep [40-50 mm]</td>
<td>9 14</td>
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</table>

4 DISCUSSION

4.1 Cable Bacteria Density and Activity in Mudflats of the Auray Estuary

Oxygen and pH microprofiles recorded at stations 2 and 3 showed the typical fingerprint of CBA: a pH maximum within the oxic zone without oxygen production followed by a significant acidification into the suboxic zone (Figure 4; Nielsen et al., 2010; Pfeffer et al., 2012; Risgaard-Petersen et al., 2012; Meysman et al., 2015). The presence of CB at station 2 was further confirmed by the qPCR data. At station 2, the 16S CB copy number was constant within the oxic and suboxic zones (upper first centimetre; Figure 4). The calculated filament density of about 7 m.cm\(^{-3}\) at this station was in the same order of magnitude than the in situ densities reported from the Baltic sea (Marzocchi et al., 2018; Hermans et al., 2019), from bivalve reefs (Malkin et al., 2017), subtidal mudflats (van de Velde et al., 2016) or intertidal salt marshes (Larsen et al., 2015). The geochemical signature at station 1 is less clear regarding Cable Bacteria Activity (CBA). There was no pH peak in the oxic zone and only a moderate acidification within the suboxic zone (\(\Delta \text{pH} < 0.1\)). This acidification was lower than expected if only driven by sulphate reduction and too high to be explained by iron reduction (van Cappellen and Wang, 1996; Soetaert et al., 2007). Therefore, we suggested that it was driven by low CBA. The qPCR data indicated very low CB filament density about 7 m.cm\(^{-3}\). The filament density was in the low range of the in situ densities reported from the Baltic sea (Marzocchi et al., 2018; Hermans et al., 2019). The diversity of pH microprofiles observed between the three
stations could indicate a contrasted intensity of the CBA between stations. According to the low filament abundance and the low range of pH ($\Delta$pH = 1.0) at station 1, the CBA would be minimal and it would have limited impact on the sediment geochemistry. Conversely, the strong abundance and pH range ($\Delta$pH = 2.4) suggest the most intense CBA at station 2, whereas pH range ($\Delta$pH = 1.8) at station 3 suggests an intermediate to high CBA.

Currently, the control factors of spatial and temporal variability of the CB density and the CBA on mudflats are still unresolved. Our observations suggest that Ulvae mats observed at stations 2 and 3 during core sampling could play a role on CB development. Several studies showed that macrophyte decay is rather slow compared to mirophytobenthic mineralization and favours free-sulphide production and upward diffusion (Anschutz et al., 2007; Metzger et al., 2007; Cesbron et al., 2014; Delgard et al., 2016). Previous observations confirm the rather high spatial and temporal CBA dynamics already mentioned in the literature (e.g. Seitaj et al., 2015; Lipsewers et al., 2017; Hermans et al., 2019).

4.2 Impact of Cable Bacteria on Living Foraminifera

The CBA causes pH anomalies that impact sediment geochemistry and lead to the carbonate dissolution process as described in Risgaard-Petersen et al. (2012), Meysmann et al. (2015), van der Velde et al. (2016) and Rao et al. (2016). By analogy, it has been supposed that CBA could also be responsible for foraminifera dissolution (Risgaard-Petersen et al., 2012; Richirt et al., 2022). We showed in Figure 4 and Figure 6 that advanced dissolution stages 3, 4 and 5 were significantly overrepresented at stations 2 and 3, where acidification was important, compared to station 1 where no DS-5 was observed. More precisely, vertical DS distribution corresponded to vertical acidity variability at stations 2 ($0.01 < \text{DS-5/C} < 0.50$) and 3 ($0.00 < \text{DS-5/C} < 0.79$). There is no indication for such depth distribution at station 1 where pH variability was the lowest ($\text{DS-5/C} = 0$). The relative abundance of calcareous specimens over agglutinated (C/T) is very stable along depth at station 1 ($0.78 \pm 0.07$; Appendix) whereas this ratio is more variable at stations 2 and 3 ($0.65 \pm 0.17$ and $0.73 \pm 0.15$ respectively), confirming that pH conditions affect assemblage composition through the under representation of calcareous foraminifera although species diversity is never affected (Figure 5). Assuming that acidification intensity in the suboxic zone is due to cable bacteria activity, our data suggest that CB have a drastic effect on the integrity of shells from living benthic foraminifera and potentially on their assemblages. The magnitude of this effect may depend in the CBA intensity and duration throughout the life cycle of foraminifera.

Since the dataset of the present study is rather limited, one can examine literature data that provides together oxygen and pH microprofiles with sub-centimetre vertical distribution of living foraminifera in intertidal mudflats first and other benthic environments then. Geochemical
Data from an intertidal mudflat of the Arcachon basin in the French Atlantic coast suggest CBA in May 2011 at N station (Cesbron et al., 2016) with a ΔpH = 1.6 and a pH minimum of 6.2 well below the oxic zone at 20-mm depth. At the same station in July 2011, all calcareous benthic foraminifera specimens showed a fully dissolved test with the organic lining remaining (DS-5/C = 1). The assemblage also showed that, *Eggerella scabra*, an agglutinated species, strongly dominated the foraminiferal assemblage at all depths down to 50 mm, except for the 0 to 5 mm layer (C/T = 0.88 ± 0.02 for the uppermost layer; C/T = 0 below). The authors assumed that test dissolution resulted from a strong acidification of the sediments due to an intense remineralisation of the relict roots of *Zostera*. We can assume here that these roots provided the refractory material that enhanced sulphate reduction (Anschutz et al., 2007; Metzger et al., 2007; Cesbron et al., 2014; Delgard et al., 2016), providing enough free-sulphide to favour CB development that drove the dissolution process as it probably happened at stations 2 and 3 of the Auray estuary in the present study. However, Cesbron and co-workers also showed that during winter (February 2011), foraminifera showed less dissolution due to a lower intensity of diagenetic processes including free-sulphide production and probably benthic acidification. These results underline the importance of the temporal variability of diagenetic processes that influence pore water geochemistry including CBA (Seitaj et al., 2015; Lipsewers et al., 2017; Hermans et al., 2019; Malkin et al., 2022), and eventually calcareous test integrity. It also questions about time integration of pH conditions recorded in the foraminifera tests as foraminifera may have mechanisms to buffer pH variations as suggested by different studies (de Nooijer et al., 2009b, 2014; Toyofuku et al., 2017) or vertical migration strategies (Geslin et al., 2004; Pucci et al., 2009; Koho et al., 2011; Hess et al., 2013). It could be assumed that the dissolution of the calcareous foraminifera tests would respond to an integrated dynamics over several months of exposure to the acidity caused by CBA, rather than immediately. These dynamics should be investigated in the future in Auray estuary to better understand differences of dissolution stages observed between stations. It can also be assumed that tolerance to acidification may be species-dependent and needs detailed investigation.

Conversely, a tidal mudflat from another estuarine system of French Atlantic coast seems not to show indices of CBA nor occurrence of dissolution on living foraminifera. Living foraminifera from the Brilliante mudflat of Loire estuary was studied at two stations in September 2012 and April 2013 (Thibault de Chanvalon et al., 2015, 2022). The vertical distribution of living foraminifera reported in the Loire mudflat was similar to the vertical distribution of station 2 reported in the present study with a maximal density at the topmost layer within the oxic zone, a minimal density around 10-mm depth and a second maximum below. However, no foraminiferal test dissolution was reported by Thibault de Chanvalon and co-workers and the foraminiferal assemblages were heavily dominated by calcareous
foraminiferal species, resulting in a DS-5/C ratio equal to zero and a C/T ratio about 1. Furthermore, at these stations, pH profiles did not show signs of CBA at different occasions (May 2013, February 2014, June 2018, unpublished data). pH decrease corresponded to oxygen uptake and was below 0.5 units with a minimum about 7.7. No pH peak at the interface was observed in a profile performed in the dark neither. The major difference between these systems is the size of the river that induces significant resuspension-deposition event for the Loire estuary limiting the development of favourable conditions to CB development. In addition, bioturbation seems to be intense at the Brillantes mudflat (Thibault de Chanvalon et al., 2015, 2016b, 2017). Another difference between these studies is the absence of macrophytes at the studied stations of the Brillantes mudflat. Finally the size of the catchment area of Loire provides an important flux of suspended particles rich in metallic oxides that will once settled in the mudflat generate a thick layer of sediment where the iron cycle dominates diagenetic processes acting as an efficient “iron curtain” that maintains free-sulphide between 5 to 10 cm depth (Thibault de Chanvalon et al., 2016b, 2016a) to be out of reach for CB. These combined conditions are not favourable to CB development (Malkin et al., 2014, 2017). This, foraminiferal observations strongly suggest the absence of CBA in the studied part of the Brillantes mudflat. This area may be considered as a control station.

Other studies reporting calcareous test dissolution of benthic living foraminifera in comparable environments are published but without geochemical data, allowing to discuss potential CBA (Alve and Nagy, 1986; Buzas-Stephens, 2005; Polovodova and Schonfeld, 2008; Bentov et al., 2009; de Nooijer et al., 2009; Kurtarkar et al., 2011; Haynert et al., 2012; Schönfeld and Mendes, 2022). Although the hypotheses put forward by these authors on the causes of test dissolution are all plausible (environmental pollution, freshwater inputs, organic matter degradation), they are not strongly explained. Therefore, the absence of pH data (Buzas-Stephens, 2005; Polovodova and Schonfeld, 2008) or its insufficient vertical resolution (Alve and Nagy, 1986; Haynert et al., 2012; Schönfeld and Mendes, 2022) does not exclude the potential involvement of CB in those environments. In the Baltic sea, that could be considered as a sort of giant estuary, Charrieau et al. (2018a), provide pH microprofiles that seem to indicate the absence of CBA. However, these authors observed calcareous test dissolution of living foraminifera and concluded that dissolution may be the consequence of a complex set of environmental factors whose ecological equilibrium can change rapidly in such coastal areas (salinity, oxygen concentration, pH and Ω_{calc}). Laboratory experiments conducted by these authors (Charrieau et al., 2018c), seem to indicate that low salinity may be an important factor on calcareous test dissolution. The difference with estuarine studies discussed above is probably that salinity change dynamics in the Baltic is rather minor compared to
salinity in Auray and Loire that are macrotidal systems with species adapted to such salinity variations.

Considering the increase of observations of cable bacteria activity occurrence in a wide range of marine environments (Malkin et al., 2014; Burdorf et al., 2017) like estuaries, coastal lagoons, salt marshes, marine lakes, tidal and subtidal mudflats, we assume that the impact of CBA on foraminifera community should be strongly considered.

4.3 Impact of Cable Bacteria on Dead Assemblages

Richirt et al. (2022) have assumed that calcareous test dissolution resulting from CBA could be responsible for low densities of calcareous tests in the dead assemblages recorded in sediments of Lake Grevelingen. Our results suggest that acidification caused by CBA strongly affects the calcareous shell integrity and the assemblage composition of living foraminifera before taphonomic processes. Our study also suggests that after foraminifera death, CBA keeps transforming the foraminifera assemblage during test burial confirming the hypothesis formulated by Richirt and coworkers (2022). Comparing C/T and DS-5/C ratios between living and dead assemblages at different depths we relate in detail the impact of pH distribution and therefore CBA to the taphonomic loss. Under a weak CBA (like at station 1), calcareous tests were relatively well preserved. At this station, the community structure between living and dead assemblages varied slightly (C/T ranged from 0.81 to 0.98 in living assemblage and from 0.74 to 0.89 in dead assemblage). The occurrence of dissolution in the living assemblage was nil while in the dead assemblage the DS-5/C ratio increased with depth from 0.06 to 0.18 indicating that the low dissolution generated a relatively slow taphonomic process. Calcareous tests dominated both living and dead assemblages with an increase of this trend with depth in the dead assemblage confirming the good preservation of calcareous foraminifera. Where CBA was moderate (like at station 3), the dissolution effect on the thanatocoenosis was gradual with depth. Calcareous test density decreased through the wide acidic layer (C/T decrease from above 0.8 to 0.36 at 50-mm depth) and there was an accumulation of fully dissolved tests showing only their organic linings in dead foraminiferal assemblages at depth (DS-5/C of 0.95). This feature suggests that the moderate dissolution generated a gradual taphonomic process leading to a noticeable calcareous loss with depth. Eventually, under a strong and intense CBA (like at station 2), the dissolution effect occurred mostly within the restricted acidic layer. The calcareous tests disappeared from the dead foraminiferal assemblage in this subsurface layer while the fully dissolved tests showing only their organic linings and agglutinated tests accumulated (C/T = 0.37 and DS-5/C = 0.96). At depth, the dead foraminiferal assemblage showed fairly high densities that are comparable to stations where CBA was less intense. As the living specimens were quite rare, such accumulation of dead tests suggested that somehow they bypassed the acidic firewall of the suboxic layer. If tests arrived at depth through
sedimentary burial the acidic firewall was possibly not active in a recent past. If CBA is not recent, physical or biological reworking buried sufficiently fast to preserve tests from corrosive conditions and mechanic crumbling. At this stage, these hypotheses cannot be assessed. One can note the high concentration of dead fully dissolved tests in the first 2 mm (0.70) where pH is the most alkaline suggesting that sedimentary reworking may have brought dead specimens from the subsurface acidic layer to the surface. Further studies on dead assemblages are needed to statistically validate the CBA vs. calcareous test loss relationship.

With low pH in pore water, the dissolution process resulting from CBA suggests an imprint on taphonomy and on historical records yet to be explored. Dissolution of living and dead calcareous test foraminifera due to CBA may be taken into account in interpretations of foraminiferal assemblages in historical studies. As proposed in Richirt et al. (2022), historical records of benthic foraminifera could also be used to reconstruct past CBA and determine the age of the first CB occurrence in the studied environments. Therefore, associating it with major environmental changes, light could be shed on the original factors of this bacterial spreading discovered only ten years ago.

5 CONCLUSION

This original study strongly suggests that sediment acidification caused by CBA could be responsible for significant calcareous test foraminifera dissolution patterns. As a result, proportions of calcareous test would change in both living and dead assemblages. The proportion of fully dissolved tests showing only their organic linings would increase in the living assemblages in the suboxic and anoxic zones of the sediment, as well as in the thanatocoenosis. The spatial dynamics of calcareous test dissolution in mudflats described in the present study seems to be the consequence of CBA which leads to a wide range of pore water pH in the suboxic zone of sediment.

Now that we have an idea of the potential impact of this bacterial activity on foraminiferal assemblages and on calcareous test preservation, we are entitled to ask what implications this might have for environmental interpretations of data from their use as paleoproxies, or bioindicators. In order to better understand this impact, it would be relevant to explore in situ and in vitro the effect of CBA at several time scales on the resilience of foraminiferal communities to learn more about the integration of their response in the historic/fossil record. Eventually, we should be able to provide a historical retrospective on the presence of CB in marine sediments and their impact on the benthic meiofauna. In this perspective, we should combine such studies with the development of biomarkers of these chemolithoautotrophic bacteria or of ancient eDNA.
Data availability. All of the data are published within this paper and in the Supplement. The raw data used to make the figures are available on request.

Author contributions. Maxime DAVIRAY (foraminiferal and geochemical analysis, writing, review and editing), Emmanuelle GESLIN (head of CB-For CNRS project, foraminiferal analysis, writing, review and editing), Nils RISGAARD-PETERSEN (statistical inference, writing, review and editing), Vincent SCHOLZ (qPCR proceeding, review and editing), Marie FOUET (field work, review and editing), Edouard METZGER (microprofile acquisition, writing, review and editing).

Competing interests. At least one of the (co-)authors is a member of the editorial board of Biogeosciences.

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<th>Ammonia sp.</th>
<th>Elphidium sp.</th>
<th>Quinqueloculina sp.</th>
<th>DS-5 specimens</th>
<th>Ammobaculites agglutinans</th>
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Appendix. Foraminiferal absolute densities and ratios in the Auray estuary for the three stations.
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<th>Size Range</th>
<th>Mean Volume</th>
<th>Actual Volume</th>
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