Running title: Biological sedimentary iron oxides

Subject: Geomicrobiology and microbial contributions to geochemical cycles

A biological source of marine sedimentary iron oxides

Jacob P. Beam[1*], Jarrod J. Scott[1], Sean M. McAllister[2], Clara S. Chan[2], James McManus[1], Filip J. R. Meysman[2], and David Emerson[1]

[1]Bigelow Laboratory for Ocean Sciences, East Boothbay, Maine, USA, 04544

[2]University of Delaware, Department of Geological Sciences, Newark, Delaware, USA, 19716

[3]Royal Netherlands Institute for Sea Research, Department of Ecosystem Studies, Korringaweg 7, 4401 NT Yerseke, The Netherlands

[*]To whom correspondence should be addressed: jbeam@bigelow.org; PO Box 380, East Boothbay, ME, USA, 04544; phone: 207-315-2567 extension 406

Abstract

The biogeochemical cycle of iron is intricately linked to numerous element cycles. Although reductive biological processes that bridge the iron cycle to other element cycles are established, little is known about microbial oxidative processes on iron cycling in sedimentary environments—resulting in the formation of iron oxides. Here, we show that a major source of sedimentary iron oxides originates from the metabolic activity of iron-oxidizing bacteria from the class Zetaproteobacteria, stimulated by burrowing animals in coastal sediments. Zetaproteobacteria were estimated to be a global total of 10^{26} cells in coastal, bioturbated sediments and would equate to an annual production of approximately 7.9 x 10^{15} grams of sedimentary iron oxides—twenty-five times larger than the annual flux of iron oxides by rivers. These data suggest that iron-oxidizing Zetaproteobacteria are keystone organisms in marine sedimentary environments given their low numerical abundance; yet exert a profound impact via the production of iron oxides.

Keywords: iron-oxidizing bacteria/Zetaproteobacteria/bioturbation/iron biogeochemistry/keystone organism

Main

Iron oxides are important components of coastal and continental shelf sediments, and are thought to originate primarily by river deposition (Poulton and Raiswell, 2002). Authigenic iron oxides can also be formed by the oxidation of ferrous iron [Fe(II)], which has been largely attributed to chemical oxidation of Fe(II) in pore waters in areas with significant sediment mixing and irrigation by animals—bioturbation and bioirrigation—and subsequent reaction with oxygen in anoxic sediments (Aller, 1982; Canfield, 1989). Although sedimentary chemical oxidation of iron is important under saturated oxygen conditions at neutral pH (Canfield, 1989), bioirrigated sediments contain microenvironments—formed by burrowing animals—that are well below saturation (Kristensen and Kostka, 2005) where the biological contribution to iron oxidation is quantitatively more significant (Emerson *et al.*, 2010). Under low-oxygen conditions (< 100 µM O_2) and without rapid replenishment of highly reactive, poorly-crystalline iron oxides (i.e., ferrihydrite and lepidocrocite), they would be quickly exhausted by hydrogen sulfide and by bacterial iron reduction, and form iron sulfides or be released to the water column. Collectively, these findings suggest that biology is involved and important in marine sedimentary iron oxidation under ferruginous conditions commonly observed in bioturbated sediments.

The Zetaproteobacteria represent a class of iron-oxidizing bacteria (FeOB) that are exclusively found in marine or saline-influenced environments that contain high ferrous iron [Fe(II)] concentrations (McAllister *et al.*, 2011; McBeth *et al.*, 2013; Scott *et al.*, 2015, 2017). Coastal marine sediments can have Fe(II) pore water concentrations ranging from ~1-2,000 µmol L^{-1} that are capable of supporting lithoautotrophic populations of Zetaproteobacteria (Emerson, 2016). Recent studies have identified Zetaproteobacteria in surface openings of benthic macrofauna in the Mediterranean Sea (Rubin-Blum *et al.*, 2014), worm burrows in sub-marine

groundwater discharge into sands in Delaware (McAllister *et al.*, 2015), coastal sediments in Denmark (Laufer *et al.*, 2016), and Baltic and North Sea sediments (Reyes *et al.*, 2016). These recent studies suggest that Zetaproteobacteria may play a significant role in iron oxidation in marine sediments—a quantitative estimate of their abundance is necessary to determine their biogeochemical role on a global scale.

We analyzed the coastal sediment microbial communities to determine the extent of Zetaproteobacteria from geographically diverse sites (n=90; Supplementary Table 1) utilizing 16S rRNA gene sequencing, which highlights their importance on a global scale. Iron oxidation appears ubiquitous in the Zetaproteobacteria (Field *et al.*, 2015; Scott *et al.*, 2015; Barco *et al.*, 2015), thus the 16S rRNA gene can be used to infer this specific metabolism. We also enriched for environmentally relevant iron-oxidizing bacteria from coastal sediments, which provided further metabolic evidence of the importance of iron oxidation in marine sediments. A meta-analysis of 16S rRNA gene studies revealed the extent and importance of Zetaproteobacteria on the global sedimentary iron biogeochemical cycle.

We identified Zetaproteobacteria in 60 % of our samples (Supplementary Table S1), and their median relative abundance was 1.1 percent of the total microbial community (range = 0.04-15 %) in worm (e.g., polychaetes) burrows in coastal marine sediments (Fig. 1a). Zetaproteobacteria were ten times less abundant in bulk sediments (Fig. 1a) with a median near zero percent (range = 0-1 %), and were statistically different from worm burrows (p-value=9.2 x 10^{-7}, Wilcoxon test). The large ranges (0.04-15 %) and non-normal distribution of Zetaproteobacteria relative abundance in worm burrows (see Supplementary Materials and Methods) was most likely a combination of differences in burrow ventilation rates and efficiencies (Kristensen and Kostka, 2005), differences in sediment physicochemical conditions

(Supplementary Table S1), and sampling bias (for example, residual sediment on worm burrows). Bioirrigation by benthic animals increases the extent of oxidative processes in these sediments, thus biological iron oxidation can occur at greater depths (10s of centimeters) than typical oxygen penetration of a few millimeters into coastal surface sediments. The abundance of Zetaproteobacteria at the burrow walls correlated with the concentration of pore water ferrous iron [Fe(II)] (Fig. 1b), which is their main energy source—resulting in the production of solid phase iron oxides around worm burrows (Fig. 1c). Quantitatively, highly reactive, iron oxides—operationally extractable by sodium dithionite (Poulton and Canfield, 2005)—were 3 times higher at burrow walls, which accounted for 20-40 % of the iron oxides with depth (Supplementary Figure S1 and S2). These freshly-produced iron oxides are important substrates for iron-reducing microorganisms that release Fe(II) into pore waters, and are essential to the supply of dissolved iron (dFe) to phytoplankton in the water column in coastal communities and continental shelves (Severmann *et al.*, 2010). Iron oxides are also important to the mineralization of organic matter in marine sediments by iron reducers (Canfield, 1989) substrates for early pyrite diagenesis (Berner, 1984), enhance organic matter burial (Lalonde *et al.*, 2012), and inhibit accumulation of pore water hydrogen sulfide, preventing conditions detrimental to benthic animals (Kristensen and Kostka, 2005), perhaps functioning as a local firewall against euxinic conditions(Seitaj *et al.*, 2015).

The relative abundance of Zetaproteobacteria in worm burrows resembles the Fe(II) concentration profile (Supplementary Figure S3), and both were at their maximum values around 2-3 cm. The high Fe(II) (~40-140 µM) and low oxygen (~20-60 µM) conditions present in bioturbated sediment pore waters (Supplementary Figure S3) were ideal habitats for microaerophilic Zetaproteobacteria to thrive (Emerson *et al.*, 2010). Zetaproteobacteria relative

abundance decreased with depth in both burrows and sediments (Supplementary Fig. S3), likely due to the decrease in Fe(II) with depth and increase in hydrogen sulfide production by sulfate-reducing bacteria. Although there is oxygen in these sediments at depth, hydrogen sulfide may inhibit oxygen respiratory machinery under these conditions. The formation of iron sulfide minerals with increasing depth by biogenic sulfide may also compete with Zetaproteobacteria for access to Fe(II). Under these ferruginous settings, biotic rates of Fe(II) oxidation exceed abiotic chemical oxidation (Emerson *et al.*, 2010).

Two Zetaproteobacteria Operational Taxonomic Units (herein, referred to as ZetaOTUs) dominated the Zetaproteobacterial diversity in worm burrows (Supplementary Table S2). The dominant ZetaOTU across all samples was ZetaOTU14, which comprised 32 % of all ZetaOTUs (Supplementary Table S2), and is represented by four single cell amplified genomes (SAGs) from diffuse flow vent systems (Field *et al.*, 2015; Scott *et al.*, 2015, 2017). We isolated the first member of ZetaOTU14, strain CSS-1 from iron oxide surface flocculent in a laboratory bioturbation microcosm (Supplementary Figure S4). This strain grew best under low oxygen (~60 µM O_2) and high Fe(II) concentrations similar to those measured from sediment pore waters (Supplementary Figure S3). Strain CSS-1 produced stalks encrusted with poorly-crystalline iron oxides under laboratory conditions (Supplementary Figure S4). These iron oxides are consistent with those produced by other Zetaproteobacteria (Chan *et al.*, 2010), as well as in naturally occurring iron mats associated with hydrothermal vents, which are highly reactive, and resistant to undergoing diagenesis to more crystalline oxides (e.g., goethite) (Picard *et al.*, 2015). Single cell genomes from ZetaOTU14 representatives contained genes essential for growth on iron and low oxygen conditions (Supplementary Table S2). The second most abundant OTU was ZetaOTU9 (22 %) and is represented by two cultured isolates (*Ghiorsea bivora* strains TAG-1

and SV108) (Mori *et al*.,, 2017), as well as 5 SAGs from deep-sea vents (Supplementary Table S2). ZetaOTU9 isolates also had genes necessary for growth on iron and low oxygen (Supplementary Table S2), and have also been shown to oxidize hydrogen, which may explain the ubiquity of this OTU in sediments and other environments (see below). There was no clear distribution of ZetaOTUs 14 and 9 with respect to depth (Supplementary Figure S3) in worm burrows and sediments—although it is likely that they are adapted for specific Fe(II) and O_2 concentrations, which were hypothesized for other Zetaproteobacteria (Field *et al.*, 2015).

We searched for Zetaproteobacterial 16S rRNA gene sequences in marine sediment datasets (Supplementary Table S3), and identified them in numerous sediments on a global scale (Figure 2). We found a pattern consistent with our samples—ZetaOTUs 14 and 9 were present and generally the most abundant ZetaOTUs in coastal and shelf sediments (Figure 2). Zetaproteobacteria relative abundance was not found to exceed one percent in other studies, as microenvironments were not considered, which are abundant in bioturbated sediments (Kristensen and Kostka, 2005). Accordingly, we hypothesize that when the abundance of Zetaproteobacteria exceeds ~0.1 % in sediments, there is active growth and iron oxidation associated with bioturbating and bioirrigating animals. We estimated a median global population size of Zetaproteobacteria to be 1.05×10^{26} cells (range = 3.83×10^{24}-1.44×10^{27} cells) from our measurements (Fig. 1a) and from other studies (Fig. 2) in continental shelf sediments using cellular abundance in the upper 10 centimeters—the worldwide average depth of bioturbation (Boudreau, 1998)—of continental shelf environments (total cells =10^{29}; <150 m water depth (Kallmeyer *et al.*, 2012)). The global Zetaproteobacteria abundance estimate was then used with recent iron oxide production rate measurements from diffuse flow vents (~1.3×10^{-16} mol Fe cell^{-1} hr^{-1}) (Emerson *et al.*, 2016), which could result in the production of ~7 petagrams of iron

159 oxides per year (range = 0.1-70 petagrams per year). Recent two-dimensional, sub-millimeter
160 Fe(II) measurements in bioturbated sediments revealed extensive Fe(II) oxidation occurring
161 within the immediate vicinity of worm burrows and a rapid re-oxidation rate of 3.78 ±1.4 mmol
162 Fe m^{-2} day^{-1} (de Chanvalon *et al.*, 2017). These chemical rate measurements combined with an
163 estimate of the global volume of bioturbated coastal sediments 10 cm deep (~2.1 x 10^{13} m^3)
164 (Teal *et al.*, 2008) would equate to an annual production of 1.6 ± 1.1 petagrams of iron oxides.
165 These two independent estimations of iron oxide production rates are well within the range of
166 one another. Based on these estimates, the annual biological oxidation of iron in sediments—
167 forming iron oxides—could exceed the annual flux of iron oxides from rivers to coastal
168 sediments (Poulton and Raiswell, 2002) up to a factor of twenty-five.
169 Zetaproteobacteria exert a profound impact on global sedimentary biogeochemistry via
170 the production of biogenic, highly-reactive iron oxides despite their low global abundance (~0.11
171 %)—effectively functioning as keystone organisms in coastal sediments stimulated by burrowing
172 animals. Zetaproteobacteria contribute significantly to the rapid rates of Fe(II) re-oxidation
173 measured and observed in coastal sediments (Chanvalon *et al.*, 2017). Climate change outcomes
174 such as coastal hypoxia may have positive or negative effects on the sedimentary iron
175 biogeochemical cycle—either stimulating microaerobic bacterial iron oxidation resulting in an
176 increase in iron oxide production, thus enhancing dFe release or inhibiting oxidation by the
177 increase in hydrogen sulfide production, precipitating Fe as iron sulfides. The result of an
178 increase or decrease in dFe flux would be enhanced or reduced primary productivity by
179 phytoplankton, respectively. Thus, sedimentary iron oxide formation by Zetaproteobacteria may
180 have a direct impact on important water column processes such as carbon and nitrogen fixation.
181

Acknowledgements

This project was funded by a National Science Foundation Biological Oceanography Award number OCE-1459600 (JPB and DE). Sample collection for the Oregon margin and Gulf of Mexico was funded by National Science Foundation grants OCE-1029889 and OCE-1147407 (to JM). We appreciate Sara George for field and laboratory assistance, Peter Larsen for marine polychaete and hemichordate identification, Anton Tramper for sampling worm burrow and sediments from Netherlands, Peter Girguis and David Johnston for helpful discussions, Megan Harder for assistance with iron oxide and DNA extractions, and Matthew Wade for logistical support.

The authors declare no conflict of interest.

References

Aller RC. (1982). The Effects of Macrobenthos on Chemical Properties of Marine Sediment and Overlying Water. In: McCall PL, Tevesz MJS (eds). *Animal-Sediment Relations: The Biogenic Alteration of Sediments*. Springer US: Boston, MA, pp 53–102.

Barco RA, Emerson D, Sylvan JB, Orcutt BN, Jacobson Meyers ME, Ramírez GA, *et al.* (2015). New insight into microbial iron oxidation as revealed by the proteomic profile of an obligate iron-oxidizing chemolithoautotroph. *Appl Environ Microbiol* **81**: 5927–5937.

Berner A. (1984). Sedimentary pyrite formation: An update. *Geochim Cosmochim Acta*

Boudreau BP. (1998). Mean mixed depth of sediments: The wherefore and the why. *Limnol Oceanogr* **43**: 524–526.

Canfield DE. (1989). Reactive iron in maine sediments. *Geochim Cosmochim Acta* **53**: 619–632.

Chan CS, Fakra SC, Emerson D, Fleming EJ, Edwards KJ. (2010). Lithotrophic iron-oxidizing

205 bacteria produce organic stalks to control mineral growth: implications for biosignature
206 formation. *ISME J* **5**: 717–727.
207 de Chanvalon AT, Metzger E, Mouret A, Knoery J, Geslin E, Meysman FJR. (2017). Two
208 dimensional mapping of iron release in marine sediments at submillimetre scale. *Mar Chem*.
209 **191**: 34-49.
210 Emerson D. (2016). The irony of iron - Biogenic iron oxides as an iron source to the ocean.
211 *Front Microbiol* **6**: 1–6.
212 Emerson D, Fleming EJ, McBeth JM. (2010). Iron-oxidizing bacteria: an environmental and
213 genomic perspective. *Annu Rev Microbiol* **64**: 561–583.
214 Emerson D, Scott JJ, Leavitt AH, Fleming E, Moyer CL. (2016). In situ estimates of iron-
215 oxidation and accretion rates for iron-oxidizing bacterial mats at Loihi Seamount. *bioRxiv*.
216 Field EK, Sczyrba A, Lyman AE, Harris CC, Woyke T, Stepanauskas R, *et al.* (2015). Genomic
217 insights into the uncultivated marine Zetaproteobacteria at Loihi Seamount. *ISME J* **9**: 857–870.
218 Kallmeyer J, Pockalny R, Adhikari RR, Smith DC, D'Hondt S. (2012). Global distribution of
219 microbial abundance and biomass in subseafloor sediment. *Proc Natl Acad Sci* **109**: 16213–
220 16216.
221 Kristensen E, Kostka JE. (2005). Macrofaunal Burrows and Irrigation in Marine Sediment:
222 Microbiological and Biogeochemical Interactions. In: Kristensen E (ed) *Interact Between*
223 *Macro- Microorg Mar Sediments*. American Geophysical Union Washington, DC, pp. 125–157.
224 Lalonde K, Mucci A, Ouellet A, Gelinas Y. (2012). Preservation of organic matter in sediments
225 promoted by iron. *Nature* **483**: 198–200.
226 Laufer K, Nordhoff M, Schmidt C, Behrens S, Jørgensen BB, Kappler A. (2016). Co-existence
227 of microaerophilic, nitrate-reducing, and phototrophic Fe(II)-oxidizers and Fe(III)-reducers in

coastal marine sediment. *Appl Environ Microbiol* **82**: 1433–1447.

McAllister SM, Barnett JM, Heiss JW, Findlay AJ, MacDonald DJ, Dow CL, *et al.* (2015). Dynamic hydrologic and biogeochemical processes drive microbially enhanced iron and sulfur cycling within the intertidal mixing zone of a beach aquifer. *Limnol Oceanogr* **60**: 329–345.

McAllister SM, Davis RE, McBeth JM, Tebo BM, Emerson D, Moyer CL. (2011). Biodiversity and emerging biogeography of the neutrophilic iron-oxidizing Zetaproteobacteria. *Appl Environ Microbiol* **77**: 5445–5457.

McBeth JM, Fleming EJ, Emerson D. (2013). The transition from freshwater to marine iron-oxidizing bacterial lineages along a salinity gradient on the Sheepscot River, Maine, USA. *Environ Microbiol Rep* **5**: 453–463.

Mori JF, Scott JJ, Hager KW, Moyer CL, Küsel K, Emerson D (2017). Physiological and ecological implications of an iron- and hydrogen-oxidizing member of the Zetaproteobacteria, Ghiorsea bivora, gen. nov. sp. nov. Accepted.

Picard A, Kappler A, Schmid G, Quaroni L, Obst M. (2015). Experimental diagenesis of organo-mineral structures formed by microaerophilic Fe(II)-oxidizing bacteria. *Nat Commun* **6**: 6277.

Poulton SW, Canfield DE. (2005). Development of a sequential extraction procedure for iron: Implications for iron partitioning in continentally derived particulates. *Chem Geol* **214**: 209–221.

Poulton SW, Raiswell R. (2002). The low-temperature geochemical cycle of iron: From continental fluxes to marine sediment deposition. *Am J Sci* **302**: 774–805.

Reyes C, Dellwig O, Dahnke K, Gehre M, Noriega-Ortega BE, Bottcher ME, *et al.* (2016). Bacterial communities potentially involved in iron-cycling in Baltic Sea and North Sea sediments revealed by pyrosequencing. *FEMS Microbiol Ecol* **92**.

Rubin-Blum M, Antler G, Tsadok R, Shemesh E, Austin JA, Coleman DF, *et al.* (2014). First

evidence for the presence of iron oxidizing zetaproteobacteria at the levantine continental margins. *PLoS One* **9**: 1–10.

Scott JJ, Breier JA, Luther GW, Emerson D. (2015). Microbial iron mats at the mid-atlantic ridge and evidence that zetaproteobacteria may be restricted to iron-oxidizing marine systems. *PLoS One* **10**: 1–19.

Scott JJ, Glazer BT, Emerson D. (2017). Bringing microbial diversity into focus: high-resolution analysis of iron mats from the Lō‘ihi Seamount. *Environ Microbiol* **19**: 301-316.

Seitaj D, Schauer R, Sulu-Gambari F, Hidalgo-Martinez S, Malkin SY, Burdorf LDW, *et al.* (2015). Cable bacteria generate a firewall against euxinia in seasonally hypoxic basins. *Proc Natl Acad Sci U S A* **112**: 13278-13283.

Severmann S, McManus J, Berelson WM, Hammond DE. (2010). The continental shelf benthic iron flux and its isotope composition. *Geochim Cosmochim Acta* **74**: 3984–4004.

Teal LR, Bulling MT, Parker ER, Solan M. (2008). Global patterns of bioturbation intensity and mixed depth of marine soft sediments. *Aquat Biol* **2**: 207–218.

Figure Legends

Figure 1. Boxplots of the relative abundance of Zetaproteobacteria (**a**) in iron-oxide lined worm burrow walls (n=29) and surrounding sediments (n=61). Notches are representative of 95 % confidence interval and the medians (solid black lines) between worm burrows and sediments (1.1 % and 0 %, respectively) are statistically different (p-value=9.2 x 10^{-7}, Wilcoxon test). Filled circles represent individual data points and open circles indicate outliers. Zetaproteobacteria relative abundance (%) as a function of pore water ferrous iron [Fe(II)] concentration (µmol L^{-1}) (**b**) from worm burrows (blue circles, fitted blue line, orange fill = 95 %

274 confidence interval) and sediments (black circles, fitted black line, grey fill = 95 % confidence
275 interval) (see Methods for details on line fits). Characteristic iron oxide lined worm burrow walls
276 (c) from "The Eddy", Sheepscot River, Maine, USA (image from 27 August 2015). Burrow
277 walls are likely created by the polychaete, *Nereis diversicolor* or hemichordate, *Saccoglossus*
278 *kowalevskii,* which are both common to these intertidal sediments in Maine.
279 **Figure 2.** Global distribution of Zetaproteobacteria in marine sediments (circles) and non-
280 sediment sites (triangles) such as hydrothermal vents. The relative abundance of
281 Zetaproteobacteria in sediments from other 16S rRNA gene studies was never above 1 % and
282 was typically within the range measured from bulk marine sediments from Maine. Sequences are
283 from numerous studies (Supplementary Table S3) that include Sanger, 454, and Illumina
284 sequencing technologies.

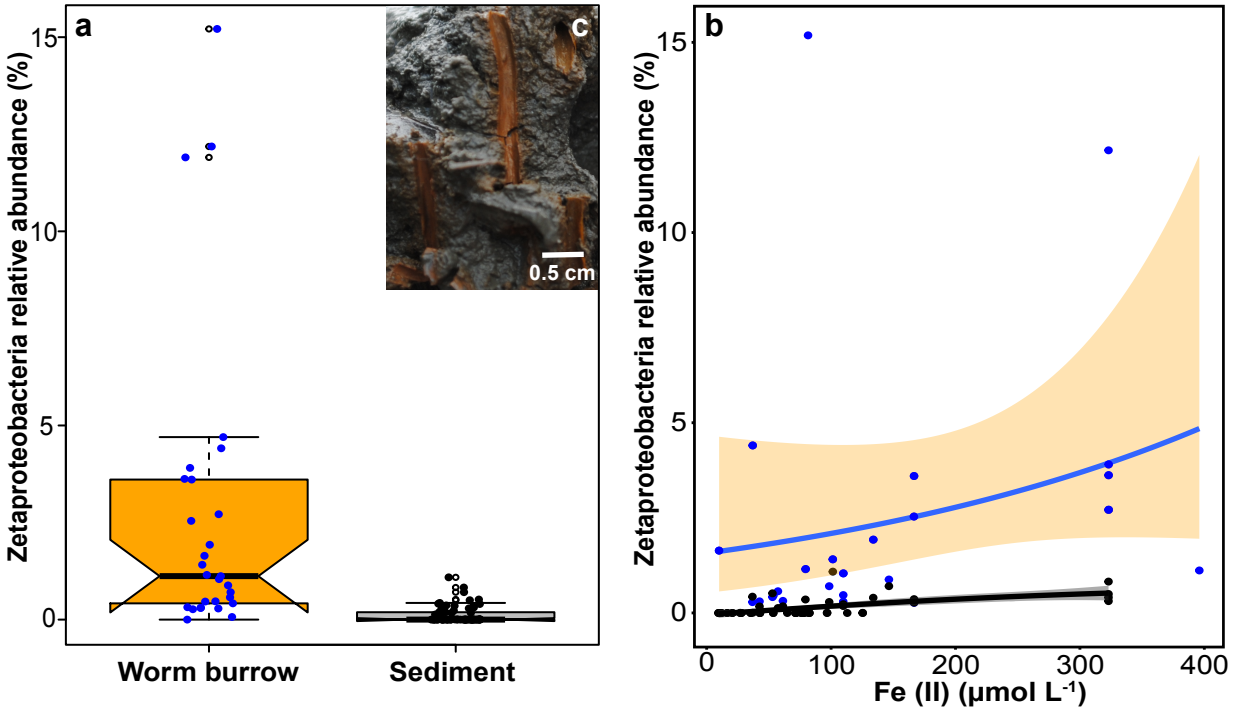

Figure 1

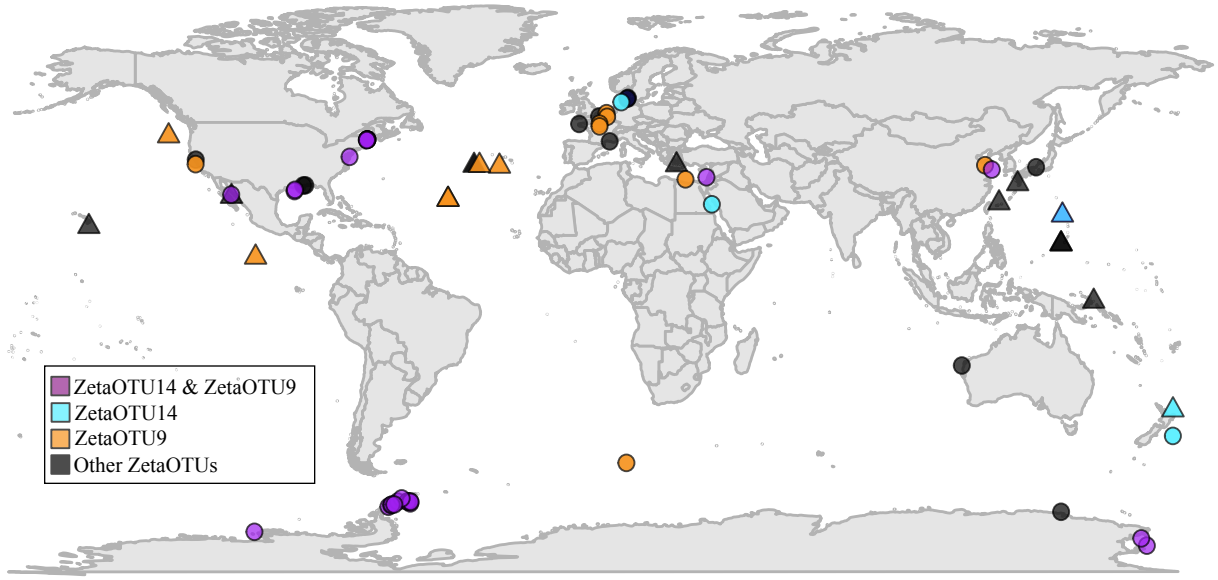

Figure 2