Non-cyanobacterial diazotrophs dominate dinitrogen fixation in biological soil crusts at the early stage of crust formation.

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1 ABSTRACT

Biological soil crusts (BSC) cover a vast global area and are key components of ecosystem productivity in arid soils. In particular, BSC contribute significantly to the nitrogen (N) budget in arid ecosystems via N2-fixation. N2-fixation in mature crusts is largely attributed to heterocystous cyanobacteria, however, early successional crusts are dominated by non-heterocystous cyanobacteria and this suggests that microorganisms other than cyanobacteria mediate N2-fixation during the early stages of BSC development. DNA stable isotope probing (DNA-SIP) with $^{15}N_2$ revealed that *Clostridiaceae* and *Proteobacteria* are the most common microorganisms to assimilate ^{15}N in early successional 'light' crusts. Incorporation of $^{15}N_2$ by cyanobateria was not observed through cyanobacteria were dominant in the crust sample. The maximum relative abundance of $^{15}N_2$-assimilating taxa in the BSC was 0.00225% and 0.00127% for taxa that belong to *Clostridiaceae* and *Proteobacteria*, respectively. Their low abundance in the BSC may explain why these heterotrophic diazotrophs have not previously been characterized in BSC. Diazotrophs play a critical role in BSC formation and characterization of these organisms represents a crucial step towards understanding how antropogenic change will effect the formation and ecological function of BSC in arid ecosystems.

2 INTRODUCTION

Biological soil crusts (BSC) are specialized microbial mat communitites that form at the soil surface in arid environmets and fill a variety of important ecological functions in arid ecosystems. BSC occupy plant interspaces and cover a wide, global geographic range (Garcia-Pichel et al., 2003b). The ground cover of BSC on the Colorado Plateau has been measured as high as 80% by remote sensing (Karnieli et al., 2003). The global biomass of BSC *Cyanobacteria* alone is estimated at 54 x 10^{12} g C (Garcia-Pichel et al., 2003b). BSC play important roles in arid ecosystem productivity and are responsible for significant nitrogen (N) flux (for review of BSC N-fixation see Belnap (2003)). For example, N-input via N-fixation versus atmospheric depositon was predominant in five times as many BSC samples from North America, Africa and Australia (Evans and Belnap, 1999). The presence of BSC is positively correlated with vascular plant survival due in part to BSC ecosystem N contributions (for review of BSC-vascular plant interactions see Belnap et al. (2003)). Climate change and disturbance alter BSC microbial community structure and membership and therefore can alter diazotroph diversity and the BSC N-budget.

28   BSC N-fixation rate studies (typically employing the acetylene reduction assay (ARA)) have explored
29 BSC diazotroph activity across various ecological gradients. Reported BSC N-fixation rates vary
30 significantly (Evans and Lange, 2001). The reasons for this variability are complex and likely include
31 the spatial heterogeneity of BSC (Evans and Lange, 2001) and the impact of recent environmental
32 conditions on N-fixation rates (see Belnap (2001) for discussion). Moreover, the ARA assay is subject
33 to methodological artifacts that preclude cross-study and possibly intra-study but inter-environment type
34 comparisons (see Belnap (2001) for review). Nonetheless, mature BSC N-fixation rate measurements
35 have been higher than younger, developing BSC N-fixation rate measurements (Belnap, 2002; Yeager
36 et al., 2004). This difference may be due to the proliferation of heterocystous *Cyanobacteria* in older mats
37 and is consistent with the theory that heterocystous *Cyanobacteria* are the primary BSC diazotrophs.
38 Alternatively, the N-fixation rate differences between young and old BSC might be attributable to
39 methodological artifacts. For instance, Johnson et al. (2005) show that N-fixation rates peak at a lower
40 depth in developing BSC as compared to mature BSC. When N-fixation is measured from intact cores of
41 developing BSC the measurement may be artifactually low due to delayed acetylene/ethylene diffusion
42 through the crust to and from the peak N-fixation rate depth in a typical ARA incubation timeframe.
43 Diffusion would not be an issue when measuring N-fixation rates in mature crust as nitrogenase activity
44 peaks near the surface. When total N-fixation rates were calculated by integrating rates over 1-3 mm depth
45 slices along full BSC cores (thus mitigating ethene/acetylene flux limitations), N-fixation rate differences
46 between developing and mature BSC were not statistically significant (Johnson et al., 2005).

47   Molecular studies of BSC microbial diversity include explorations of the BSC microbial community
48 vertical profile (Garcia-Pichel et al., 2003a), BSC *nifH* gene content surveys (e.g. Yeager et al. (2004),
49 Yeager et al. (2012), Yeager et al. (2006) and Steppe et al. (1996)), and next-generation-sequencing (NGS)
50 enabled studies of BSC SSU rRNA gene content across wide geographic ranges (Garcia-Pichel et al.,
51 2013; Steven et al., 2013). *nifH* surveys have been conducted across BSC development stages (Yeager
52 et al., 2004), as well as across seasons, temperatures and precipitation gradients (Yeager et al., 2012).
53 Mature, more fully developed BSC possess greater numbers of heterocystous *Cyanobacteria* (e.g. *Nostoc,*
54 *Syctonema*) than developing BSC but both young and old BSC are dominated by non-heterocystous
55 *Cyanobacteria* (*Microcoleus vaginatus* or *M. steenstrupii*) (Yeager et al., 2004; Garcia-Pichel et al., 2013).
56 Young or recently disturbed BSC are often described as "light" in appearance relative to "dark" mature
57 BSC (Belnap, 2002; Yeager et al., 2004). Heterocystous *Cyanobacteria* are the numerically dominant
58 BSC diazotrophs in *nifH* clone libraries (Yeager et al., 2006, 2004, 2012) although an early survey of
59 Colorado Plateau BSC *nifH* diversity recovered *nifH* genes related to *Gammaproteobacteria* as well
60 as a clade that included *nifH* genes from the anaerobes *Clostridium pssteurianum*, *Desulfovibrio gigas*
61 and *Chromatium buderi*, Specifically, Yeager et al. (2006)–in a study of overall BSC *nifH* diversity–
62 categorized 89% of 693 *nifH* sequences derived from Colorado Plateau and New Mexico BSC samples
63 as heterocystous cyanobacterial (non-cyanobacterial *nifH* sequences were largely attributed to alpha-
64 and beta- *proteobacteria*). The heterocystous cyanobacterial BSC diazotrophs fall into three genera,
65 *Scytonema*, *Spirirestis*, and *Nostoc* (Yeager et al., 2006, 2012).

66   The influence of microbial community membership and structure on BSC N-fixation is an ongoing
67 research question (Belnap, 2013). While the presence/abundance of heterocystous *Cyanobacteria* has
68 been proposed as the mechanism behind increased N-fixation in mature BSC, it is unclear if mature BSC
69 actually fix more N (see Johnson et al. (2005)). More studies are necessary to elucidate the microbial
70 membership influence on BSC N-fixation and to determine if heterocystous *Cyanobacteria* are the only
71 keystone diazotrophs. The first step in defining structure function relationships with respect to N-fixation
72 is a full accounting of BSC diazotrophs. Towards this end we conducted $^{15}N_2$ DNA stable isotope probing
73 (DNA-SIP) experiments with light, developing Colorado Plateau BSC. DNA-SIP with $^{15}N_2$ has not been
74 attempted with BSC. DNA-SIP would provides an accounting of *active* diazotrophs whereas *nifH* clone
75 libraries account for microbes with the genomic potential for N-fixation. Further, we track the distribution
76 of putative diazotrophs uncovered in this study through collections of NGS SSU rRNA libraries from BSC
77 microbial diversity surveys over a range of spatial scales and soil types (Garcia-Pichel et al., 2013; Steven
78 et al., 2013).

3 RESULTS

3.1 ORDINATION OF CSCL GRADIENT FRACTION SSU RRNA LIBRARIES

79 BSC were incubated for 4 days in the presence or absence of $^{15}N_2$ and DNA was extracted for DNA-
80 SIP at 2 and 4 days. Fractionation of CsCl gradients permitted separation of DNA on the basis of
81 buoyant density. Ordination of Bray-Curtis (Bray and Curtis, 1957) distances beteen SSU-rRNA amplicon
82 sequence collections from gradient fractions reveals that labeled gradient fraction (i.e. gradient fractions
83 of DNA from $^{15}N_2$ incubations) sequence collections diverge from control (i.e. DNA from incubations
84 without $^{15}N_2$) at the "heavy" of the CsCl gradients (Figure S2). Although the density position of gradient
85 fractions from different gradients do not match perfectly, fraction pairs from corresponding control versus
86 labeled gradients can be constructed by pairing control gradient fractions with their closest density
87 neighbors/fractions from corresponding labeled gradients. If a gradient fraction did not have a mate
88 within a density difference of 0.003 g/mL it remained unpaired. Bray-Curtis distance between the fraction
89 pairs is positively correlated to the density of the labeled fraction (p-value: 0.00052, r^2: 0.3315) (inset
90 Figure S2). Additionally, differences among label/control groups with heavy fractions are statistically
91 significant by the Adonis test (p-value: 0.001, r^2: 0.136) (Anderson, 2001). The first principal axis appears
92 to be correlated with fraction density (Figure S2) and the Adonis test p-value for density versus pairwise
93 Bray-Curtis distances with all CsCl fraction libraries is 0.001 (r^2 0.117).

3.2 IDENTITIES OF OTUS RESPONSIVE TO $^{15}N_2$

94 A statistically significant increase in OTU abundance in heavy fractions of $^{15}N_2$ labeled samples relative
95 to corresponding gradient fractions from controls provides evidence for OTUs that have incorporated
96 ^{15}N into their DNA. Specifically, we compared OTU proportion means between labeled and control
97 samples from heavy gradient fractions using statistics developed to find differentially expressed genes with
98 RNASeq data (CITE McMurdie and DESeq2). p-values were adjusted by the BH method CITE and we
99 used a false discovery rate (FDR) cutoff of 0.10 (typical FDR threshold in gene expression data analysis,
100 CITE DESeq2) to reject the null hypothesis that labeled versus control proportion mean differences were
101 below a chosen threshold (see methods). With the above methods 38 OTUs had labeled versus control
102 proportion mean difference p-values below 0.10 for one or both incubation days. These OTUs likely
103 to incorporated ^{15}N into DNA ($^{15}N_2$ "responders"). Of these 38, 26 are annotated as *Firmicutes*, 9
104 as *Proteobacteria*, 2 as *Acidobacteria* and 1 as *Actinobacteria* (Figure 1). If the OTUs are ranked by
105 descending, moderated proportion mean labeled:control ratios, the top 10 ratios (i.e. the 10 OTUs that
106 were most enriched in the labeled gradients considering only heavy fractions) are either *Firmicutes* (6
107 OTUs) or *Proteobacteria* (4 OTUs) (Figure 2). *Proteobacteria* OTU centroid sequences for the top 10
108 responders all share high identity (>98.48% identity, Table 1) with cultivars from genera known to possess
109 diazotrophs including *Klebsiella*, *Shigella*, *Acinetobacter*, and *Ideonella*. None of the *Firmicutes* OTUs
110 in the top 10 responders share greater than 97% sequence identity with sequences in the LTP database
111 (release 115) (see Table 1).

3.3 COMPARISON OF SEQUENCE COLLECTIONS AT "STUDY"-LEVEL

112 *3.3.1 Comparisons of OTU content:* There were 3,079 OTUs (209,354 total sequences after quality
113 control) in the DNA-SIP data, 3,203 OTUs (129,033 total sequences after quality control) in the Garcia-
114 Pichel et al. (2013) study, and 2,481 OTUs (129,358 total sequences after quality control) in the Steven
115 et al. (2013) study. Of the 4,340 OTU centroids established for this study (including sequences from
116 Steven et al. (2013) and Garcia-Pichel et al. (2013)) 445 have matches in the Living Tree Project (LTP)
117 (a collection of 16S gene sequences for all sequenced type strains (Yarza et al., 2008)) at greater or equal
118 than 97% (LTP version 115). That is, 445 of 4,340 are closely related to cultivars. The DNA-SIP data
119 set shares 56% OTUs with the Steven et al. (2013) data and 46% of OTUs with the Garcia-Pichel et al.

120   (2013) data (where total OTUs are from the combined data for each pairwise comparison). The Steven
121   et al. (2013) and Garcia-Pichel et al. (2013) share 46% of OTUs.

122   *3.3.2   Comparisons of Taxonomic Content:*   *Cyanobacteria* and *Proteobacteria* were the top two
123   phylum-level sequence annotations for all three studies but only the DNA-SIP data had more
124   *Proteobacteria* annotations than *Cyanobacteria*. *Proteobacteria* represented the 29.8% of sequence
125   annotations in DNA-SIP data as opposed to 17.8% and 19.2% for the Garcia-Pichel et al. (2013) and
126   Steven et al. (2013) data, respectively. There is a stark contrast in the total percentage of sequences
127   annotated as *Firmicutes* between the raw environmental samples and the DNA-SIP data. *Firmicutes*
128   represent only 0.21% and 0.23% of total phylum level sequence annotations in the Steven et al. (2013)
129   and Garcia-Pichel et al. (2013) studies, respectively (Figure S1). In the DNA-SIP sequence collection
130   *Firmicutes* make up 19% of phylum level sequence annotations. Also in sharp contrast for the DNA-
131   SIP versus environmental data is the number of putative heterocystous *Cyanobacteria* sequences. Only
132   0.29% of *Cyanobacteria* sequences in the DNA-SIP data are annotated as belonging to "Subsection IV"
133   which is the heterocystous order of *Cyanobacteria* in the Silva taxonomic nomenclature (Pruesse et al.,
134   2007). In the Steven et al. (2013) and Garcia-Pichel et al. (2013) studies 15% and 23%, respectively, of
135   *Cyanobacteria* sequences are annotated as belonging to "Subsection IV".

3.4   DISTRIBUTION OF BSC DIAZOTROPHS IN ENVIRONMENTAL SAMPLES

136   *3.4.1   Clostridiacea:*   Five of the 6 *Firmicutes* in the top 10 responder OTUs (above) belong in
137   the *Clostridiacea*. We only observed one of these strongly responding *Clostridiaceae* in the data
138   presented by Garcia-Pichel et al. (2013), "OTU.108" (closest BLAST hit in LTP Release 115 –
139   *Caloramotor proteoclasticus*, BLAST %ID 96.94, Accession X90488). OTU.108 was found in two
140   samples both characterized as "light" crust. One other *Clostridiaceae* OTU with a proportion mean ratio
141   (labeled:control) p-value less than 0.10 but outside the top 10 responders was found in the Garcia-Pichel
142   et al. (2013) data (a "light" crust sample). None of the strongly responding *Clostridiacea* were found in the
143   sequences provided by Steven et al. (2013). *Clostridiaceae* ^{15}N-responder OTU centroid 16S sequences
144   are generally more closely related to environmental than cultivar 16S gene sequences.

145   *3.4.2   Proteobacteria:*   One of the *Proteobacteria* OTUs in the 10 most strongly responding OTUs was
146   found in the Garcia-Pichel et al. (2013) sequences (closest BLAST hit in LTP Release 115, BLAST %ID
147   100, Accession ZD3440, *Acinetobacter johnsonii*). None of the strongly responding *Protebacteria* OTUs
148   were found in the Steven et al. (2013) sequences. There were 133 responder OTU-sample occurrences
149   (responder OTU was found in a sample library) in the Steven et al. (2013) data. 83 were in "below crust"
150   samples, 50 in BSC samples.

151   *3.4.3   Other taxa:*   Two ^{15}N-responsive OTUs were found in an extensive number of environmental
152   samples (61 of 65 samples from the combined data sets of Garcia-Pichel et al. (2013) and Steven et al.
153   (2013)). Both OTUs were annotated as *Acidobacteria* but shared little sequence identity to any cultivar
154   SSU rRNA gene sequences in the LTP (Release 115), with best LTP BLAST hits of 81.91 and 81.32%
155   identity. Additionally, the evidence for ^{15}N incorporation for each OTU was weak relative to other putative
156   responders (adjusted p-values of 0.090 and 0.096). Of the remaining 36 stable isotope responder OTUs,
157   only 14 were observed in the environmental data. Figure S5 summarizes the OTU-sample occurrences in
158   both the Steven et al. (2013) and the Garcia-Pichel et al. (2013) data with occurrences distributed into the
159   most relevant sample classes of each study.

4 DISCUSSION

4.1 STUDY-LEVEL DIFFERENCES

160 SIP places focus upon organisms based on isotope incorporation and has the ability to detect activity by
161 low abundance members of the community. DNA from OTUs that incopororate ^{15}N into their biomass
162 moves towards the heavy end of the CsCl gradient and therefore OTUs in "labeled" DNA are enriched
163 in the full data pool relative to bulk DNA. Phylum-level taxonomic annotations such as *Firmicutes* and
164 *Proteobacteria* of ^{15}N-responsive OTUs are enriched in the DNA-SIP data relative to environmental data
165 (Figure X).

4.2 ORDINATION OF CSCL GRADIENT FRACTION 16S RRNA GENE SEQUENCE COLLECTIONS

166 The ordination of Bray-Curtis distances between CsCl gradient fraction 16S sequence collections show
167 that control fractions differ from labeled fractions in the "heavy" range of the CsCl gradients (Figure S2).
168 If each control fraction is paired to the labeled fraction from the same incubation day for which it is closest
169 in density, there is a positive and statistically significant correlation between Bray-Curtis distances within
170 fraction pairs and density of the pair (see inset Figure S2). Therefore, the "heavy" end of the control
171 and labeled gradients differ and the OTUs enriched in the labeled fractions (relative to control) would
172 have incorporated ^{15}N into their DNA during the incubation timeframe. If the incubation timeframe is
173 appropriate, the ^{15}N-incorporators would most likely have incorporated the ^{15}N from atmospheric ^{15}N$_2$.

4.3 BSC DIAZOTROPHS IDENTIFIED IN THE STUDY

174 BSC N-fixation has long been attributed to heterocystous *Cyanobacteria* and molecular microbial ecology
175 surveys of BSC *nifH* gene content have been consistent with this hypothesis finding cyanobacterial *nifH*
176 types to be numerically dominant in *nifH* gene libraries (Yeager et al., 2006, 2004, 2012). Even poorly
177 developed BSC samples have yielded predominantly cyanobacterial *nifH* genes (Yeager et al., 2004).
178 And, "sub-biocrust" samples have yielded *entirely* heterocystous cyanobacterial *nifH* genes (Yeager
179 et al., 2012). It is possible, however, that PCR-driven molecular surveys of *nifH* gene content have
180 been biased against non-heterocystous *Cyanobacteria*. In general the nifH PCR primers used by Yeager
181 et al. (2006, 2004, 2012) (19F and nifH3) for the first round of nested PCR have broad specificity and
182 display at least 86% *in silico* coverage for *Proteobacteria, Cyanobacteria* and "Cluster III" *nifH* reference
183 sequences (Gaby and Buckley, 2012). In the second round of the nested PCR protocol (Yeager et al.,
184 2006, 2004, 2012), primer nifH11 is' slightly biased against "Cluster III" (50% coverage) but biased
185 in favor of *Proteobacteria* (79% *in silico* coverage against 67% for *Cyanobacteria*) and nifH22 matches
186 *Proteobacteria, Cyanobacteria* and "Cluster III" reference sequences poorly (16%, 23% and 21% *in silico*
187 coverage, respectively) (Gaby and Buckley, 2012). Unfortunately, it is difficult to assess or quantify this
188 bias (in either direction) without knowing the *nifH* gene content *de novo*. Another potential bias in favor of
189 *Cyanobacteria* in BSC *nifH* gene libraries is heterocysts (the specialized N-fixing cells along the trichome
190 of filamentous heterocystous *Cyanobacteria* such as *Nostoc* and *Scytonema*) may be overrepresented
191 with respect to non-cyanobacterial diazotrophs because heterocysts make up a fraction cells along a
192 trichome and even non-heterocyst cells in a trichome will possess the *nifH* gene. Polyploidy could further
193 exacerbate this bias, as many *Cyanobacteria* are estimated to have multiple genome copies per cell (Griese
194 et al., 2011). Moreover, it should also be noted that *nifH* gene content is not directly extrapolable to
195 the taxonomic relative abundances of nitrogenase proteins. Regardless, our results suggest that BSC N-
196 fixation may include a significant non-cyanobacterial component that requires further assessment across
197 a more comprehensive sampling of BSC types.

198 We did not observe evidence for N-fixation by heterocystous *Cyanobacteria* in the "light" crust samples
199 used in this study. One possible explanation for our results is that the "light", still developing BSC samples

used in this study possessed too few heterocystous *Cyanobacteria* to statistically evaluate their ^{15}N-incorporation. Indeed, only 0.29% of sequences from this study's DNA-SIP 16S rRNA gene sequence libraries were from heterocystous *Cyanobacteria* (see results) as opposed to 15% and 23% of total sequences in the Steven et al. (2013) and Garcia-Pichel et al. (2013) data, respectively. It is difficult to compare relative abundance values from CsCl gradient fractions against environmental libraries, but, a three order of magnitude difference between the environmental libraries and the CsCl gradient fractions is stark. Nonetheless, we would still expect even low abundance diazotrophs to show evidence for ^{15}N-incorporation, provided sequence counts were not too sparse in heavy fractions. The OTUs defined by selected heterocystous *Cyanobacteria* sequences presented in Yeager et al. (2006), however, all fall below the sparsity threshold used in our analysis (see methods, Figure **??**). Given the sparsity of heterocystous *Cyanobacteria* sequences in the DNA-SIP data set, it is not possible to assess whether heterocystous *Cyanobacteria* incorporated ^{15}N during the incubation. It should be noted that "light" and in particular "sub-biocrust" samples possess much less heterocystous *Cyanobacteria* in general (Figure S3) so the samples used in this study are not necessarily unrepresentative of typical poorly developed BSC simply because they are lacking heterocystous *Cyanobacteria*.

The OTUs that did appear to incorporate ^{15}N during the incubation were predominantly *Proteobacteria* and *Firmicutes*. The *Proteobacteria* OTUs for which ^{15}N-incorporation signal was strongest all shared high sequence identity ($\geq$98.48% sequence identity) with 16S sequences from cultivars in genera with known diazotrophs (Table 1). The *Firmicutes* that displayed signal for ^{15}N-incorporation (predominantly *Clostridiaceae*) were not closely related to any cultivars (Table 1, Figure 3). These BSC *Clostrodiaceae* diazotrophs represent a gap in culture collections. As culture-based ecophysiological studies have proven useful towards explaining ecological phenomena in BSC 16S rRNA gene sequence libraries (Garcia-Pichel et al., 2013), it would seem that these putative *Clostridiaceae* diazotrophs would be prime candidates for targeted culturing efforts. Assessing the physiological response of these diazotrophic *Clostridiaceae* to temperature would be useful for predicting how climate change will affect the BSC nitrogen budget.

This would be place to mention that C. pasteuranium was firs known diazotroph, isolated from soil by Wintogradsky. Also the place to mention cyanobacterial compatible solutes and their dynamics in relation to wetting of dry soil. This relates to spore formes with a boom-bust ecology.

Although too undersampled in the environmental data sets to reach statistical conclusions, ^{15}N-responsive OTUs were found more often in below crust samples (as opposed to BSC samples) in the Steven et al. (2013) data and in "light" BSC samples in the Garcia-Pichel et al. (2013) data (Figure S5). This result generates some hypotheses that are counter to prior discussions regarding BSC diazotroph temporal dynamics (keeping in mind this phenomenon has not been evaluated statistically). Specifically, the transition of BSC from a light colored, developing crust to a dark, mature crust may not mark the *emergence* of diazotrophs in BSC but rather the *transition* of the diazotroph community from heterotroph dominance to cyanobacterial. Additionally, the soil beneath BSC may contribute significantly to the N budget in arid ecosystems.

4.4 SEQUENCING DEPTH

Rarefaction curves of all samples from Steven et al. (2013) and Garcia-Pichel et al. (2013) are still sharply increasing especially for "below crust" samples (Figure S4). Parametric richness estimates of BSC diversity indicate the Steven et al. (2013) and Garcia-Pichel et al. (2013) sequencing efforts recovered on average 40.5% (sd. 9.99%) and 45.5% (sd. 11.6%) of existing 16S OTUs from samples (inset Figure S4), respectively. Further, the Steven et al. (2013) and Garcia-Pichel et al. (2013) sequence collections only share 57.6% of total OTUs found in at least one of the studies. In fact, this study shares more OTUs with Steven et al. (2013), 62.4% of OTUs in the combined data, than the Steven et al. (2013) study shares with Garcia-Pichel et al. (2013). Therefore, is not alarming that few of the ^{15}N-responsive OTUS were

246 found by Garcia-Pichel et al. (2013) and Steven et al. (2013), it is important to point out that even next-
247 generation sequencing efforts of BSC 16S rRNA genes have only shallowly sampled the full diversity of
248 BSC microbes.

4.5 CONCLUSION

249 Heterocystous *Cyanobacteria* are key contributors to the BSC N-budget, but, the putative diazotrophs
250 elucidated in this study and in Steppe et al. (1996) in addition to the N-fixation rate data presented by
251 (Johnson et al., 2005) suggest there may be significant non-cyanobacterial BSC diazotrophs specifically
252 within the *Clostrideaceae* and *Proteobacteria*. It seems clear that heterocystous *Cyanobacteria* increase
253 in abundance with BSC age (Yeager et al., 2004). It is less clear if this transition marks the emergence of
254 diazotrophy versus a re-structuring of the BSC diazotroph community from one dominated by *Firmicutes*
255 and *Proteobacteria* to one predominantly heterocystous *Cyanobacteria*. DNA-SIP is a valuable tool in
256 the molecular microbial ecologist's toolbox for identifying members of microbial community functional
257 guilds (Neufeld et al., 2007). PCR-based surveys of diagnostic marker genes and DNA-SIP are both used
258 to connect microbial phylogenetic types to microbial activities, but they occupy a non-overlapping set of
259 strengths and weaknesses. Combined these tools can powerfully reveal connections between ecosystem
260 membership/structure and function. Here we supplement previous surveys of BSC *nifH* diversity, a
261 diagnostic marker PCR-driven approach, with $^{15}N_2$ DNA-SIP, and, while we do not confirm previous
262 results, we expand knowledge of BSC diazotroph diversity. Predicting BSC N-fixation with respect to
263 climate change, althered precipitation regimes and physical disturbance requires a careful accounting of
264 diazotrophs including non-cyanobacterial types.

5 MATERIALS AND METHODS

5.1 BSC SAMPLING AND INCUBATION CONDITIONS

265 Light crust samples (37.5 cm^2, average mass 35 g) were incubated in sealed chambers under controlled
266 atmosphere and in the light for 4 days. Crusts were dry prior to time zero and were wetted at initiation of
267 experiment. Treatments included control air (unenriched headspace) and enriched air (>98% atom $^{15}N_2$)
268 headspace. Samples were taken at 2 days and 4 days incubation. Acetylene reduction rates were measured
269 daily. DNA was extracted from 1 g of crust. Samples were taken from Green Butte, Arizona as previously
270 described (site CP3, Beraldi-Campesi et al. (2009)). All samples were from light crusts as described by
271 Johnson et al. (2005).

5.2 DNA EXTRACTION

272 DNA from each sample was extracted using a MoBio PowerSoil DNA Isolation Kit (following
273 manufacturers protocol, but substituting a 2 minute bead beating for the vortexing step), and then gel
274 purified. Extracts were quantified using PicoGreen nucleic acid quantification dyes (Molecular Probes).

5.3 DNA-SIP

275 Gradient density centrifugation of DNA was undertaken in 4.7 mL polyallomer centrifuge tubes in a
276 TLA-110 fixed angle rotor (both Beckman Coulter) in CsCl gradients with an average density of 1.725
277 g/mL. Average density for all prepared gradients was checked with an AR200 refractometer before runs.
278 Between 2.5-5 μg of DNA extract was added to the CsCl solution, and gradients were run under conditions
279 of 20C for 67 hours at 55,000 rpm (Lueders et al., 2004) CITE. Centrifuged gradients were fractionated
280 from bottom to top in 36 equal fractions of 100 μL, using a by syringe pump as described Manefield et
281 al. (2002). The density of each fraction was determined using using an AR200 refractometer modified to

282 accomidate 5ul samples CITE. DNA in each fraction was desalted through four washes with 300 μL TE
283 per fraction.

5.4 PCR, LIBRARY NORMALIZATION AND DNA SEQUENCING

284 Barcoded PCR of bacterial and archaeal 16S rRNA genes, in preparation for 454 Pyrosequencing, was
285 carried out using primer set 515F/806R (Walters et al., 2011) (primers purchased from Integrated DNA
286 Technologies). The primer 806R contained an 8 bp barcode sequence, a "TC" linker, and a Roche 454
287 B sequencing adaptor, while the primer 515F contained the Roche 454 A sequencing adapter. Each 25
288 μL reaction contained 1x PCR Gold Buffer (Roche), 2.5 mM MgCl$_2$, 200 μM of each of the four dNTPs
289 (Promega), 0.5 mg/mL BSA (New England Biolabs), 0.3 μM of each primers, 1.25 U of Amplitaq Gold
290 (Roche), and 8 μL of template. Template for each sample was added at normalized amounts in an attempt
291 to prevent chimera formation, and each sample was amplified in triplicate. Thermal cycling occurred
292 with an initial denaturation step of 5 minutes at 95C, followed by 40 cycles of amplification (20s at
293 95C, 20s at 53C, 30s at 72C), and a final extension step of 5 min at 72C. Triplicate amplicons were
294 pooled and purified using Agencourt AMPure PCR purification beads, following manufacturers protocol.
295 Once cleaned, amplicons were quantified using PicoGreen nucleic acid quantification dyes (Molecular
296 Probes) and pooled together in equimolar amounts. Samples were sent to the Environmental Genomics
297 Core Facility at the University of South Carolina (now Selah Genomics) to be run on a Roche FLX 454
298 pyrosequencing machine.

5.5 DATA ANALYSIS

299 *5.5.1 Sequence quality control*   Sequences were initially screened by maximum expected errors at a
300 specific read length threshold (Edgar, 2013) which has been shown to be as effective as denoising 454
301 reads with respect to removing pyrosequencing errors. Specifically, reads were first truncated to 230
302 nucleotides (nt) (all reads shorter than 230 nt were discarded) and any read that exceeded a maximum
303 expected error threshold of 1.0 was removed. After truncation and max expected error trimming, 91% of
304 original reads remained. The first 30 nt representing the forward primer and barcode on high quality,
305 truncated reads were trimmed. Remaining reads were taxonomically annotated using the "UClust"
306 taxonomic annotation framework in the QIIME software package (Caporaso et al., 2010; Edgar, 2010)
307 with cluster seeds from Silva SSU rRNA database (Pruesse et al., 2007) 97% sequence identity OTUs as
308 reference (release 111Ref). Reads annotated as "Chloroplast", "Eukaryota", "Archaea", "Unassigned" or
309 "mitochondria" were culled from the dataset. Finally, reads were aligned to the Silva reference alignment
310 provided by the Mothur software package (Schloss et al., 2009) using the Mothur NAST aligner (DeSantis
311 et al., 2006). All reads that did not appear to align to the expected amplicon region of the SSU rRNA gene
312 were discarded. Quality control parameters removed 34716 of 258763 raw reads.

313 *5.5.2 Sequence clustering*   Sequences were distributed into OTUs using the UParse methodology
314 (Edgar, 2013). Specifically, cluster seeds were identified using USearch with a collection of non-redundant
315 reads sorted by count as input. The sequence identity threshold for establishing a new OTU centroid
316 was 97%. After initial cluster centroid selection, select 16S rRNA gene sequences trimmed to the same
317 alignment positions as the other centroids from Yeager et al. (2006) were added to the centroid collection.
318 Specifically, Yeager et al. (2006) Colorado Plateau or Moab, Utah sequences were added which included
319 the 16S sequences for *Calothrix* MCC-3A (accession DQ531700.1), *Nostoc commune* MCT-1 (accession
320 DQ531903), *Nostoc commune* MFG-1 (accession DQ531699.1), *Scytonema hyalinum* DC-A (accession
321 DQ531701.1), *Scytonema hyalinum* FGP-7A (accession DQ531697.1), *Spirirestis rafaelensis* LQ-10
322 (accession DQ531696.1). Centroid sequences that matched selected Yeager et al. (2006) sequences with
323 greater than to 97% sequence identity were subsequently removed from the centroid collection. With
324 USearch/UParse, potential chimeras are identified during OTU centroid selection and are not allowed to
325 become cluster centroids effectively removing chimeras from the read pool. All quality controlled reads
326 were then mapped to cluster centroids at an identity threshold of 97% again using USearch. 95.6% of

327 quality controlled reads could be mapped to centroids. Unmapped reads do not count towards sample
328 counts and are essentially removed from downstream analyses. The USearch software version for cluster
329 generation was 7.0.1090.

330 *5.5.3 Merging data from this study, Garcia-Pichel et al. (2013), and Steven et al. (2013)* As only
331 sequences without corresponding quality scores were publicly available from Garcia-Pichel et al. (2013)
332 and Steven et al. (2013), these data sets were only quality screened by determining if they covered the
333 expected region of the 16S gene (described above). All data (this study, Garcia-Pichel et al. (2013)
334 and Steven et al. (2013)) were included as input to USearch for OTU centroid selection and subsequent
335 mapping to OTU centroids.

336 *5.5.4 Phylogenetic tree* The alignment for the "*Clostridiaceae*" phylogeny was created using SSU-
337 Align which is based on Infernal (Nawrocki and Eddy, 2013; Nawrocki et al., 2009). Columns in
338 the alignment that were not included in the SSU-Align covariance models or were aligned with poor
339 confidence (less than 95% of characters in a position had posterior probability alignment scores of
340 at least 95%) were masked for phylogenetic reconstruction. Additionally, the alignment was trimmed
341 to coordinates such that all sequences in the alignment began and ended at the same positions. The
342 "*Clostridiaceae*" tree included all top BLAST hits (parameters below) for ^{15}N *Clostridiaceae* responders
343 in the Living Tree Project database (Yarza et al., 2008) in addition to BLAST hits within a sequence
344 identity threshold of 97% to ^{15}N responders from the Silva SSURef_NR SSU rRNA database (Pruesse
345 et al., 2007). Only one SSURef_NR115 hit per study per OTU ("study" was determined by "title" field)
346 was selected for the tree. FastTree (Price et al., 2010) was used to build the tree and support values are
347 SH-like scores reported by FastTree.

348 *Placement of short sequences into backbone phylogeny* Short sequences were mapped to the reference
349 backbone using pplacer (Matsen et al., 2010) (default parameters). pplacer finds the edge placements that
350 maximize phylogenetic likelihood. Prior to being mapped to the reference tree, short sequences were
351 aligned to the reference alignment using Infernal (Nawrocki et al., 2009) against the same SSU-Align
352 covariance model used to align reference sequences.

353 *5.5.5 BLAST searches* BLAST searches were done with the "blastn" program from BLAST+ toolkit
354 (Camacho et al., 2009) version 2.2.29+. Default parameters were always employed and the BioPython
355 (Cock et al., 2009) BLAST+ wrapper was used to invoke the blastn program. Pandas (McKinney, 2012)
356 and dplyr (Wickham and Francois, 2014) were used to parse and munge BLAST output tables.

357 *5.5.6 Identifying OTUs that incorporated ^{15}N into their DNA* SIP is a culture-independent approach
358 towards defining identity-function connections in microbial communities (Buckley, 2011; Neufeld et al.,
359 2007). Microbes incubated in the presence of ^{13}C or ^{15}N labeled substrates can incorporate the stable
360 heavy isotope into biomass if they participate in the substrate's transformation. Stable isotope labeled
361 nucleic acids can then be separated from unlabeled by buoyant density in a CsCl gradient. As the buoyant
362 density of a macromolecule is dependent on many factors in addition to stable isotope incorporation
363 (e.g. GC-content in nucleic acids (Youngblut and Buckley, 2014)), labeled nucleic acids from one
364 microbial population may have the same buoyant density of unlabeled nucleic acids from another (i.e.
365 each population's nucleic acids would be found at the same point along a density gradient although
366 only one population's nucleic acids are labeled). Therefore it is imperative to compare density gradients
367 with nucleic acids from heavy stable isotope incubations to gradients from "control" incubations where
368 everything mimics the experimental conditions except that unlabeled substrates are used (and all DNA
369 would be unlabeled). By contrasting "heavy" density gradient fractions in experimental density gradients
370 (hereafter referred to as "labeled" gradients) against heavy fractions in control gradients, the identities of
371 microbes with labeled nucleic acids can be determined

372 We used an RNA-Seq differential expression statistical framework (Love et al., 2014) to find OTUs
373 enriched in heavy fractions of labeled gradients relative to corresponding density fractions in control
374 gradients (for review of RNA-Seq differential expression statistics applied to microbiome OTU count data
375 see McMurdie and Holmes (2014)). We use the term differential abundance (coined by McMurdie and
376 Holmes (2014)) to denote OTUs that have different proportion means across sample classes (in this case
377 the only sample class is labeled/control). CsCl gradient fractions were categorized as "heavy" or "light".
378 The heavy category denotes fractions with density values above 1.725 g/mL. Since we are only interested
379 in enriched OTUs (labeled versus control), we used a one-sided z-test for differential abundance (the null
380 hypothesis is the labeled:control proportion mean ratio for an OTU is less than a selected threshold). P-
381 values were corrected with the Benjamini and Hochberg method (Benjamini and Hochberg, 1995). We
382 selected a $\log_2$ fold change null threshold of 0.25 (or a labeled:control proportion mean ratio of 1.19).
383 DESeq2 was used to calculate the moderated $\log_2$ fold change of labeled:control proportion mean ratios
384 and corresponding standard errors. Mean ratio moderation allows for reliable ratio ranking such that
385 high variance and likely statistically insignificant mean ratios are appropriately shrunk and subsequently
386 ranked lower than they would be as raw ratios. To summarize, OTUs with high moderated labeled:control
387 proportion mean ratios have higher proportion means in heavy fractions of labeled gradients relative to
388 heavy fractions of control gradients, and therefore have likely incorporated ^{15}N into their DNA during the
389 incubation.

390 Although DNA-SIP is a powerful technique, analysis of DNA-SIP data is not without ambiguities. One
391 limitation is the discrete, selected boundary in the form of a adjusted p-value threshold (or false discovery
392 rate) that marks which OTUs we consider to be enriched in the heavy fractions of labeled CsCl gradients
393 (and thus have likely incorporated ^{15}N into their DNA during the incubation). In reality the metric we
394 use to quantify the magnitude of an OTU's response to a stable isotope is continuous, and there is only
395 an artificial boundary between which OTUs appear to have "responded" and which OTUs have unknown
396 response. For this reason, we have presented all the OTUs that satisfy our "response" criteria but focused
397 on the most strongly responding OTUs. As with any hypothesis-based statistical test, care should be taken
398 when interpreting the significance of results where p-values are near the selected threshold for rejecting
399 the null hypothesis.

400 *5.5.7 Ordination*   Principal coordinate ordinations depict the relationship between samples at each time
401 point (day 2 and 4). Bray-Curtis distances were used as the sample distance metric for ordination. The
402 Phyloseq (McMurdie and Holmes, 2014) wrapper for Vegan (Oksanen et al., 2013) (both R packages) was
403 used to compute sample values along principal coordinate axes. GGplot2 (Wickham, 2009) was used to
404 display sample points along the first and second principal axes.

405 *5.5.8 Differential abundance in environmental samples*   Significance of OTU proportion mean
406 differences with mean annual temperature (for Garcia-Pichel et al. (2013) data) and sample type ("BSC"
407 or "below crust" Steven et al. (2013) data) was determined using the DESeq2 framework (McMurdie and
408 Holmes, 2014; Love et al., 2014). A sparsity threshold of 0.40 was set to screen out sparse OTUs. No
409 p-value correction was done for differential abundance in environmental samples as only six OTUs were
410 considered for any test.

5.6 RICHNESS ANALYSES

411 Rarefaction curves were created using bioinformatics modules in the PyCogent Python package (Knight
412 et al., 2007). Parametric richness estimates were made with CatchAll using only the best model for total
413 OTU estimates (Bunge, 2010).

REFERENCES

Marti J. Anderson. A new method for non-parametric multivariate analysis of variance. *Austral Ecology*, 26(1):32–46, Feb 2001. doi: 10.1111/j.1442-9993.2001.01070.pp.x. URL http://dx.doi.org/10.1111/j.1442-9993.2001.01070.pp.x.

J. Belnap. Factors Influencing Nitrogen Fixation and Nitrogen Release in Biological Soil Crusts. In *Biological Soil Crusts: Structure Function, and Management*, pages 241–261. Springer Science + Business Media, 2001. doi: 10.1007/978-3-642-56475-8_19. URL http://dx.doi.org/10.1007/978-3-642-56475-8_19.

J. Belnap. Factors Influencing Nitrogen Fixation and Nitrogen Release in Biological Soil Crusts. In Jayne Belnap and OttoL. Lange, editors, *Biological Soil Crusts: Structure, Function, and Management*, volume 150 of *Ecological Studies*, pages 241–261. Springer Berlin Heidelberg, 2003. ISBN 978-3-540-43757-4. doi: 10.1007/978-3-642-56475-8_19. URL http://dx.doi.org/10.1007/978-3-642-56475-8_19.

J. Belnap, R. Prasse, and K.T. Harper. Influence of Biological Soil Crusts on Soil Environments and Vascular Plants. In Jayne Belnap and OttoL. Lange, editors, *Biological Soil Crusts: Structure, Function, and Management*, volume 150 of *Ecological Studies*, pages 281–300. Springer Berlin Heidelberg, 2003. ISBN 978-3-540-43757-4. doi: 10.1007/978-3-642-56475-8_21. URL http://dx.doi.org/10.1007/978-3-642-56475-8_21.

Jayne Belnap. Nitrogen fixation in biological soil crusts from southeast Utah USA. *Biology and Fertility of Soils*, 35(2):128–135, Apr 2002. doi: 10.1007/s00374-002-0452-x. URL http://dx.doi.org/10.1007/s00374-002-0452-x.

Jayne Belnap. Some Like It Hot, Some Not. *Science*, 340(6140):1533–1534, 2013. doi: 10.1126/science.1240318. URL http://www.sciencemag.org/content/340/6140/1533.short.

Yoav Benjamini and Yosef Hochberg. Controlling the False Discovery Rate: A Practical and Powerful Approach to Multiple Testing. *Journal of the Royal Statistical Society. Series B (Methodological)*, 57 (1):289–300, 1995. ISSN 00359246. doi: 10.2307/2346101. URL http://dx.doi.org/10.2307/2346101.

H. Beraldi-Campesi, H. E. Hartnett, A. Anbar, G. W. Gordon, and F. Garcia-Pichel. Effect of biological soil crusts on soil elemental concentrations: implications for biogeochemistry and as traceable biosignatures of ancient life on land. *Geobiology*, 7(3):348–359, jun 2009. doi: 10.1111/j.1472-4669.2009.00204.x. URL http://dx.doi.org/10.1111/j.1472-4669.2009.00204.x.

J. Roger Bray and J. T. Curtis. An Ordination of the Upland Forest Communities of Southern Wisconsin. *Ecological Monographs*, 27(4):325, Oct 1957. doi: 10.2307/1942268. URL http://dx.doi.org/10.2307/1942268.

Daniel H. Buckley. Stable Isotope Probing Techniques Using 15N. In *Stable Isotope Probing and Related Technologies*, pages 129–147. American Society of Microbiology, jan 2011. doi: 10.1128/9781555816896.ch7. URL http://dx.doi.org/10.1128/9781555816896.ch7.

John Bunge. Estimating the Number of Species with Catchall. In *Biocomputing 2011*, pages 121–130. WORLD SCIENTIFIC, nov 2010. doi: 10.1142/9789814335058_0014. URL http://dx.doi.org/10.1142/9789814335058_0014.

C Camacho, G Coulouris, V Avagyan, N Ma, J Papadopoulos, K Bealer, and TL Madden. BLAST+: architecture and applications. 10:421, Dec 2009.

JG Caporaso, J Kuczynski, J Stombaugh, K Bittinger, FD Bushman, EK Costello, N Fierer, AG Pea, JK Goodrich, JI Gordon, GA Huttley, ST Kelley, D Knights, JE Koenig, RE Ley, CA Lozupone, D McDonald, BD Muegge, M Pirrung, J Reeder, JR Sevinsky, PJ Turnbaugh, WA Walters, J Widmann, T Yatsunenko, J Zaneveld, and R Knight. QIIME allows analysis of high-throughput community sequencing data. 7:335–6, 2010.

PJ Cock, T Antao, JT Chang, BA Chapman, CJ Cox, A Dalke, I Friedberg, T Hamelryck, F Kauff, B Wilczynski, and Hoon MJ de. Biopython: freely available Python tools for computational molecular biology and bioinformatics. 25:1422–3, 2009.

463 TZ Jr DeSantis, P Hugenholtz, K Keller, EL Brodie, N Larsen, YM Piceno, R Phan, and GL Andersen.
464   NAST: a multiple sequence alignment server for comparative analysis of 16S rRNA genes. 34:W394–9,
465   2006.
466 RC Edgar. Search and clustering orders of magnitude faster than BLAST. 26:2460–1, 2010.
467 RC Edgar. UPARSE: highly accurate OTU sequences from microbial amplicon reads. 10:996–8, 2013.
468 R. D. Evans and J. Belnap. Long-Term Consequences of Disturbance on Nitrogen Dynamics in an Arid
469   Ecosystem. *Ecology*, 80(1):150–160, Jan 1999. doi: 10.1890/0012-9658(1999)080[0150:ltcodo]2.
470   0.co;2. URL http://dx.doi.org/10.1890/0012-9658(1999)080[0150:LTCODO]2.
471   0.CO;2.
472 R. D. Evans and O. L. Lange. Biological Soil Crusts and Ecosystem Nitrogen and Carbon Dynamics.
473   In *Biological Soil Crusts: Structure Function, and Management*, pages 263–279. Springer Science +
474   Business Media, 2001. doi: 10.1007/978-3-642-56475-8_20. URL http://dx.doi.org/10.
475   1007/978-3-642-56475-8_20.
476 John Christian Gaby and Daniel H. Buckley. A Comprehensive Evaluation of {PCR} Primers to Amplify
477   the {nifH} Gene of Nitrogenase. {PLoS} {ONE}, 7(7):e42149, jul 2012. doi: 10.1371/journal.pone.
478   0042149. URL http://dx.doi.org/10.1371/journal.pone.0042149.
479 F. Garcia-Pichel, S. L. Johnson, D. Youngkin, and J. Belnap. Small-Scale Vertical Distribution of Bacterial
480   Biomass and Diversity in Biological Soil Crusts from Arid Lands in the Colorado Plateau. *Microbial
481   Ecology*, 46(3):312–321, Nov 2003a. doi: 10.1007/s00248-003-1004-0. URL http://dx.doi.
482   org/10.1007/s00248-003-1004-0.
483 F. Garcia-Pichel, V. Loza, Y. Marusenko, P. Mateo, and R. M. Potrafka. Temperature Drives the
484   Continental-Scale Distribution of Key Microbes in Topsoil Communities. *Science*, 340(6140):
485   1574–1577, Jun 2013. doi: 10.1126/science.1236404. URL http://dx.doi.org/10.1126/
486   science.1236404.
487 Ferran Garcia-Pichel, Jayne Belnap, Susanne Neuer, and Ferdinand Schanz. Estimates of global
488   cyanobacterial biomass and its distribution. *Algological Studies*, 109(1):213–227, 2003b.
489 Marco Griese, Christian Lange, and Jrg Soppa. Ploidy in cyanobacteria. *FEMS Microbiology Letters*, 323
490   (2):124–131, sep 2011. doi: 10.1111/j.1574-6968.2011.02368.x. URL http://dx.doi.org/10.
491   1111/j.1574-6968.2011.02368.x.
492 SL Johnson, CR Budinoff, J Belnap, and F Garcia-Pichel. Relevance of ammonium oxidation within
493   biological soil crust communities. 7:1–12, 2005.
494 A. Karnieli, R.F. Kokaly, N.E. West, and R.N. Clark. Remote Sensing of Biological Soil Crusts. In
495   Jayne Belnap and OttoL. Lange, editors, *Biological Soil Crusts: Structure, Function, and Management*,
496   volume 150 of *Ecological Studies*, pages 431–455. Springer Berlin Heidelberg, 2003. ISBN 978-
497   3-540-43757-4. doi: 10.1007/978-3-642-56475-8_31. URL http://dx.doi.org/10.1007/
498   978-3-642-56475-8_31.
499 Rob Knight, Peter Maxwell, Amanda Birmingham, Jason Carnes, J Gregory Caporaso, Brett C Easton,
500   Michael Eaton, Micah Hamady, Helen Lindsay, Zongzhi Liu, Catherine Lozupone, Daniel McDonald,
501   Michael Robeson, Raymond Sammut, Sandra Smit, Matthew J Wakefield, Jeremy Widmann, Shandy
502   Wikman, Stephanie Wilson, Hua Ying, and Gavin A Huttley. {PyCogent}: a toolkit for making sense
503   from sequence. *Genome Biol*, 8(8):R171, 2007. doi: 10.1186/gb-2007-8-8-r171. URL http://dx.
504   doi.org/10.1186/gb-2007-8-8-r171.
505 M. I. Love, W. Huber, and S. Anders. Moderated estimation of fold change and dispersion for {RNA}-
506   Seq data with {DESeq}2. Technical report, feb 2014. URL http://dx.doi.org/10.1101/
507   002832.
508 Frederick A Matsen, Robin B Kodner, and E Virginia Armbrust. pplacer: linear time maximum-likelihood
509   and Bayesian phylogenetic placement of sequences onto a fixed reference tree. *BMC Bioinformatics*,
510   11(1):538, 2010. doi: 10.1186/1471-2105-11-538. URL http://dx.doi.org/10.1186/
511   1471-2105-11-538.
512 Wes McKinney. pandas: Python Data Analysis Library. Online, 2012. URL http://pandas.
513   pydata.org/.
514 PJ McMurdie and S Holmes. Waste not, want not: why rarefying microbiome data is inadmissible. 10:
515   e1003531, 2014.

516 EP Nawrocki and SR Eddy. Infernal 1.1: 100-fold faster RNA homology searches. 29:2933–5, Nov 2013.

517 EP Nawrocki, DL Kolbe, and SR Eddy. Infernal 1.0: inference of RNA alignments. 25:1335–7, May
518   2009.

519 JD Neufeld, J Vohra, MG Dumont, T Lueders, M Manefield, MW Friedrich, and JC Murrell. DNA
520   stable-isotope probing. 2:860–6, 2007.

521 Jari Oksanen, F. Guillaume Blanchet, Roeland Kindt, Pierre Legendre, Peter R. Minchin, R. B. O'Hara,
522   Gavin L. Simpson, Peter Solymos, M. Henry H. Stevens, and Helene Wagner. *vegan: Community*
523   *Ecology Package*, 2013. URL `http://CRAN.R-project.org/package=vegan`. R package
524   version 2.0-10.

525 MN Price, PS Dehal, and AP Arkin. FastTree 2–approximately maximum-likelihood trees for large
526   alignments. 5:e9490, Mar 2010.

527 E Pruesse, C Quast, K Knittel, BM Fuchs, W Ludwig, J Peplies, and FO Glckner. SILVA: a comprehensive
528   online resource for quality checked and aligned ribosomal RNA sequence data compatible with ARB.
529   35:7188–96, 2007.

530 PD Schloss, SL Westcott, T Ryabin, JR Hall, M Hartmann, EB Hollister, RA Lesniewski, BB Oakley,
531   DH Parks, CJ Robinson, JW Sahl, B Stres, GG Thallinger, Horn DJ Van, and CF Weber.
532   Introducing mothur: open-source, platform-independent, community-supported software for describing
533   and comparing microbial communities. 75:7537–41, 2009.

534 T.F. Steppe, J.B. Olson, H.W. Paerl, R.W. Litaker, and J. Belnap. Consortial N2 fixation: a strategy
535   for meeting nitrogen requirements of marine and terrestrial cyanobacterial mats. *FEMS Microbiology*
536   *Ecology*, 21(3):149–156, Nov 1996. doi: 10.1111/j.1574-6941.1996.tb00342.x. URL `http://dx.`
537   `doi.org/10.1111/j.1574-6941.1996.tb00342.x`.

538 Blaire Steven, La Verne Gallegos-Graves, Jayne Belnap, and Cheryl R. Kuske. Dryland soil microbial
539   communities display spatial biogeographic patterns associated with soil depth and soil parent material.
540   *FEMS Microbiol Ecol*, 86(1):101–113, May 2013. doi: 10.1111/1574-6941.12143. URL `http:`
541   `//dx.doi.org/10.1111/1574-6941.12143`.

542 WA Walters, JG Caporaso, CL Lauber, D Berg-Lyons, N Fierer, and R Knight. PrimerProspector: de novo
543   design and taxonomic analysis of barcoded polymerase chain reaction primers. 27:1159–61, Apr 2011.

544 Hadley Wickham. *ggplot2: elegant graphics for data analysis*. Springer New York, 2009. ISBN 978-0-
545   387-98140-6. URL `http://had.co.nz/ggplot2/book`.

546 Hadley Wickham and Romain Francois. *dplyr: dplyr: a grammar of data manipulation*, 2014. URL
547   `http://CRAN.R-project.org/package=dplyr`. R package version 0.2.

548 Pablo Yarza, Michael Richter, Jörg Peplies, Jean Euzeby, Rudolf Amann, Karl-Heinz Schleifer, Wolfgang
549   Ludwig, Frank Oliver Glöckner, and Ramon Rosselló-Móra. The All-Species Living Tree project: A
550   16S rRNA-based phylogenetic tree of all sequenced type strains. *Systematic and Applied Microbiology*,
551   31(4):241–250, Sep 2008. doi: 10.1016/j.syapm.2008.07.001. URL `http://dx.doi.org/10.`
552   `1016/j.syapm.2008.07.001`.

553 Chris M. Yeager, Jennifer L. Kornosky, Rachael E. Morgan, Elizabeth C. Cain, Ferran Garcia-Pichel,
554   David C. Housman, Jayne Belnap, and Cheryl R. Kuske. Three distinct clades of cultured heterocystous
555   cyanobacteria constitute the dominant N2-fixing members of biological soil crusts of the Colorado
556   Plateau USA. *FEMS Microbiology Ecology*, 60(1):85–97, 2006. doi: 10.1111/j.1574-6941.2006.00265.
557   x. URL `http://dx.doi.org/10.1111/j.1574-6941.2006.00265.x`.

558 Chris M. Yeager, Cheryl R. Kuske, Travis D. Carney, Shannon L. Johnson, Lawrence O. Ticknor, and
559   Jayne Belnap. Response of Biological Soil Crust Diazotrophs to Season Altered Summer Precipitation,
560   and Year-Round Increased Temperature in an Arid Grassland of the Colorado Plateau, USA. *Front.*
561   *Microbio.*, 3, 2012. doi: 10.3389/fmicb.2012.00358. URL `http://dx.doi.org/10.3389/`
562   `fmicb.2012.00358`.

563 CM Yeager, JL Kornosky, DC Housman, EE Grote, J Belnap, and CR Kuske. Diazotrophic community
564   structure and function in two successional stages of biological soil crusts from the Colorado Plateau
565   and Chihuahuan Desert. 70:973–83, 2004.

566 ND Youngblut and DH Buckley. Intra-genomic variation in G+C content and its implications for DNA
567   stable isotope probing (DNA-SIP). Aug 2014.

6 FIGURES AND LONG TABLES
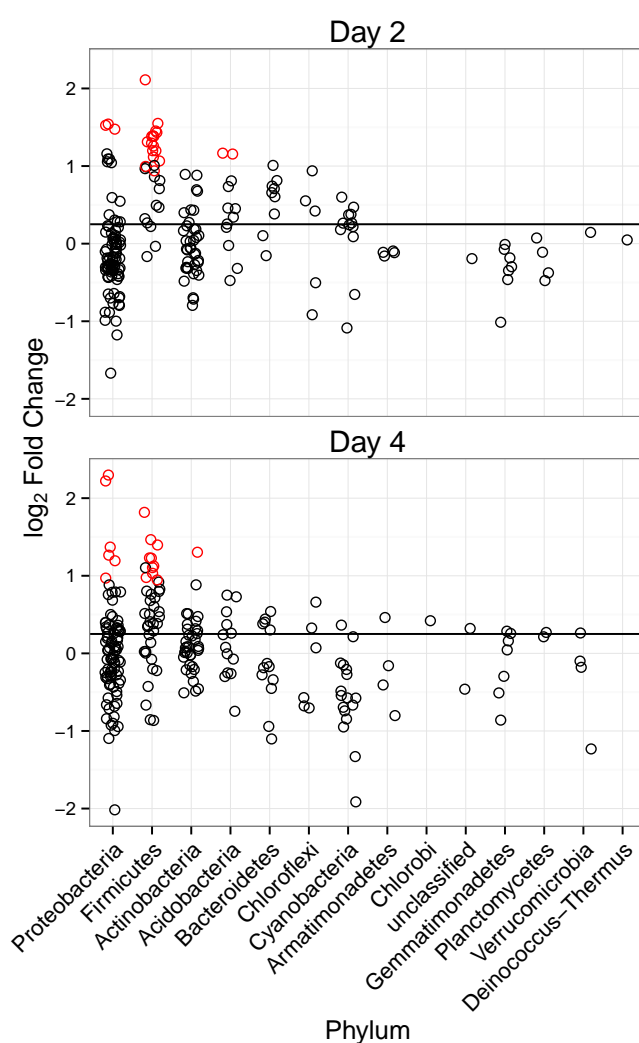
Figure 1. Moderated $\log_2$ of proportion mean ratios for labeled versus control gradients (heavy fractions only, densities ¿1.725 g/mL). All OTUs found in at least 62.5% of heavy fractions at a specific incubation day are shown. Red color denotes a proportion mean ratio that has a corresponding adjusted p-value below a false discovery rate of 10% (the null model is that the proportion mean is ratio is below 0.25). The horizontal line is the proportion mean threshold for the null model, 0.25. The inset figure summarizes the taxonomy of OTUs that with proportion mean ratio p-vaules under 0.10 for at least one time point.
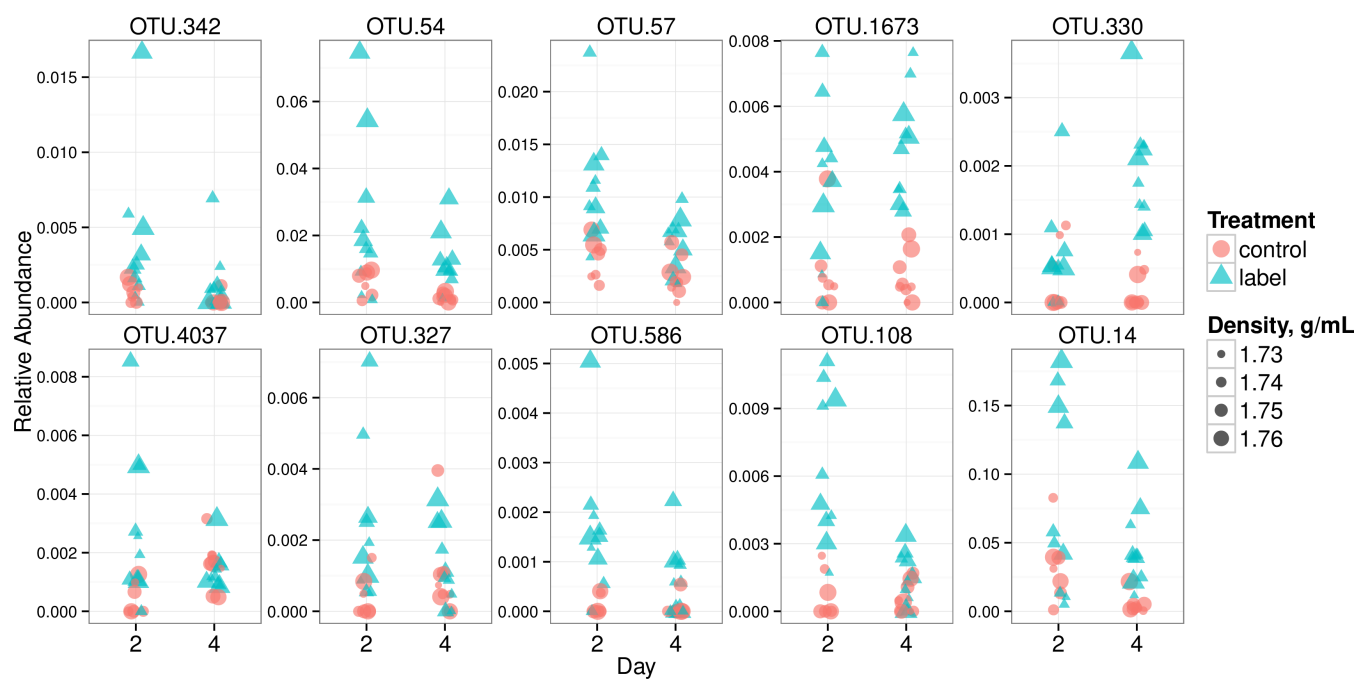
Figure 2. Relative abundance values in heavy fractions (density greater or equal to 1.725 g/mL) for the top 10 ^{15}N "responders" (putative diazotrophs, see results for selection criteria of top 10) at each incubation day. See Table 1 for BLAST results of top 10 responders against the LTP database (release 115). Point area is proportional to CsCl gradient fraction density, and color signifies control (red) or labeled (blue) treatment.
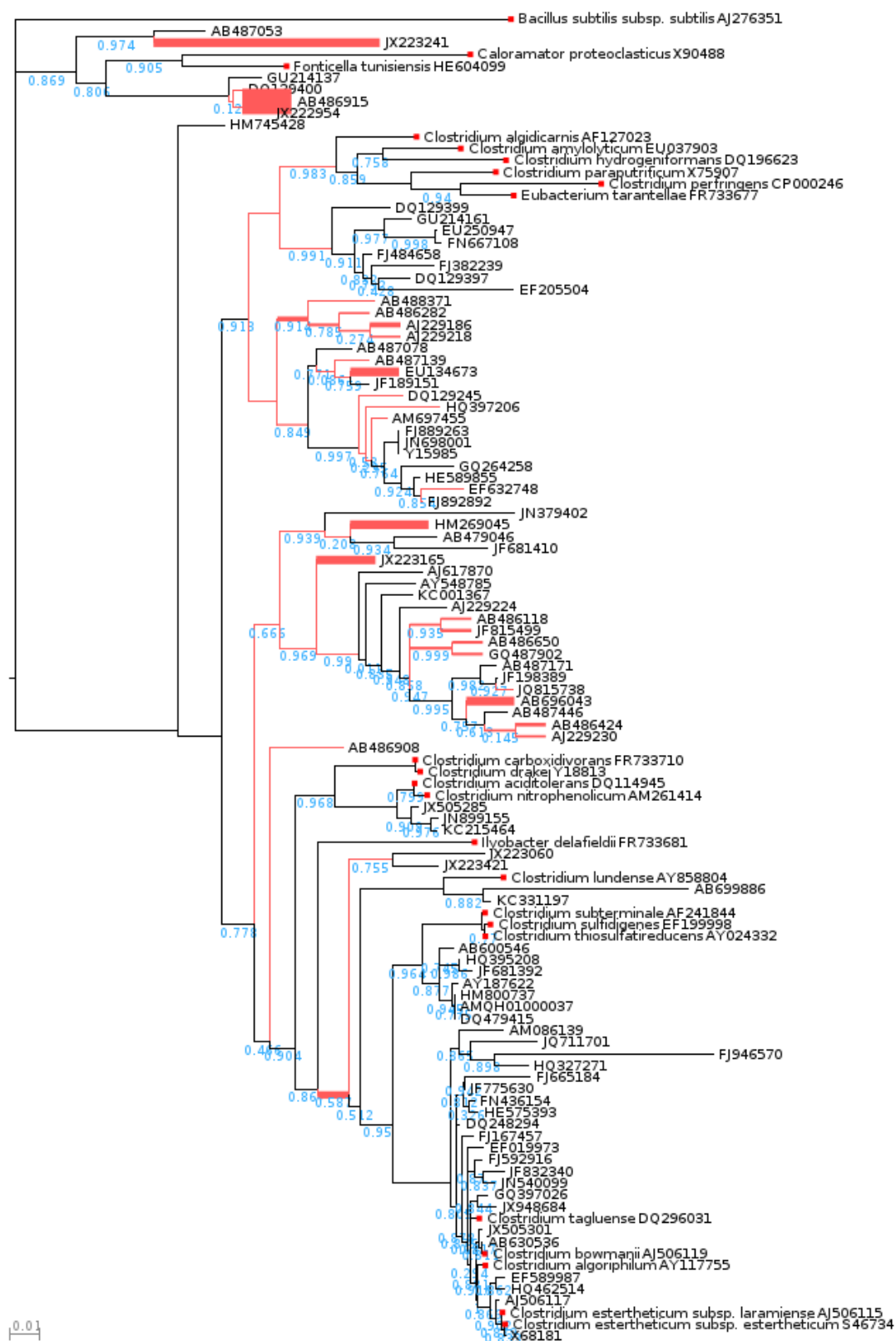
Figure 3. See methods for selection criteria for sequences in backbone tree. Edge width is proportional to number of short putative *Clostridiaceae* diazotroph sequences placed at that position. Placement of short sequences can be spread across multiple edges Matsen et al. (2010). Reference sequences from cultivars have boxes at tips and full species names. Tips with only accession annotations are from environmental reference sequences.

7   SUPPLEMENTAL FIGURES
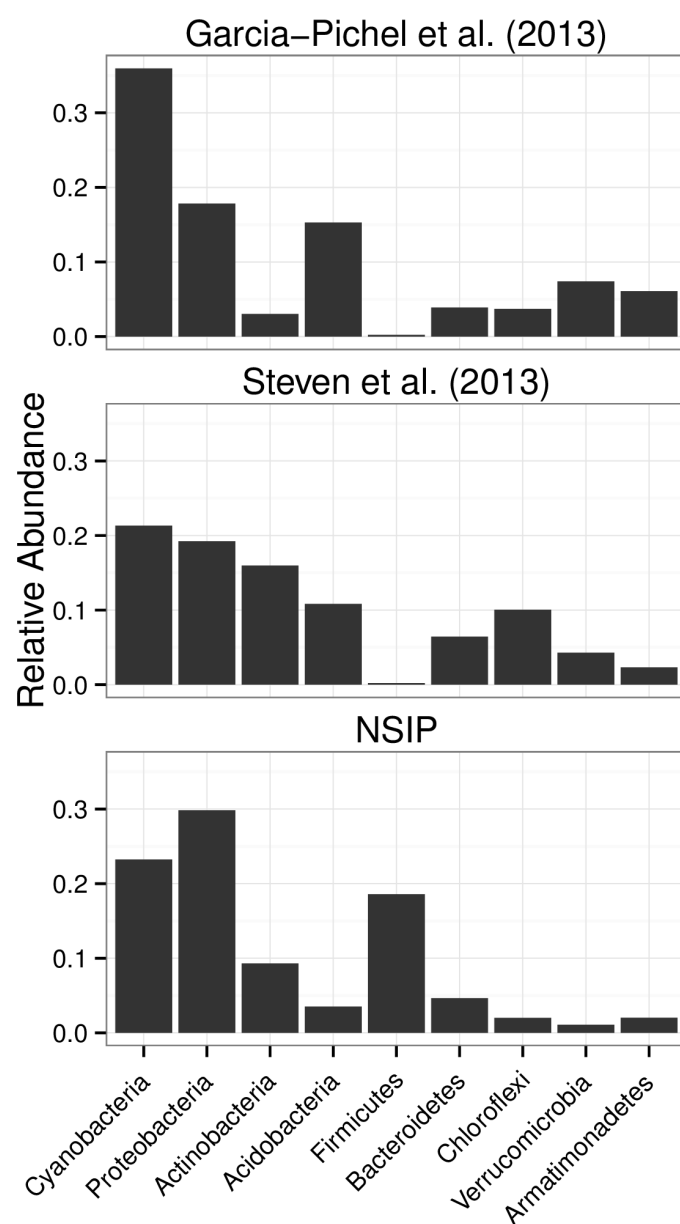
Figure S1. Distribution of sequences into top 9 phyla (phyla ranked by sum of all sequence annotations).
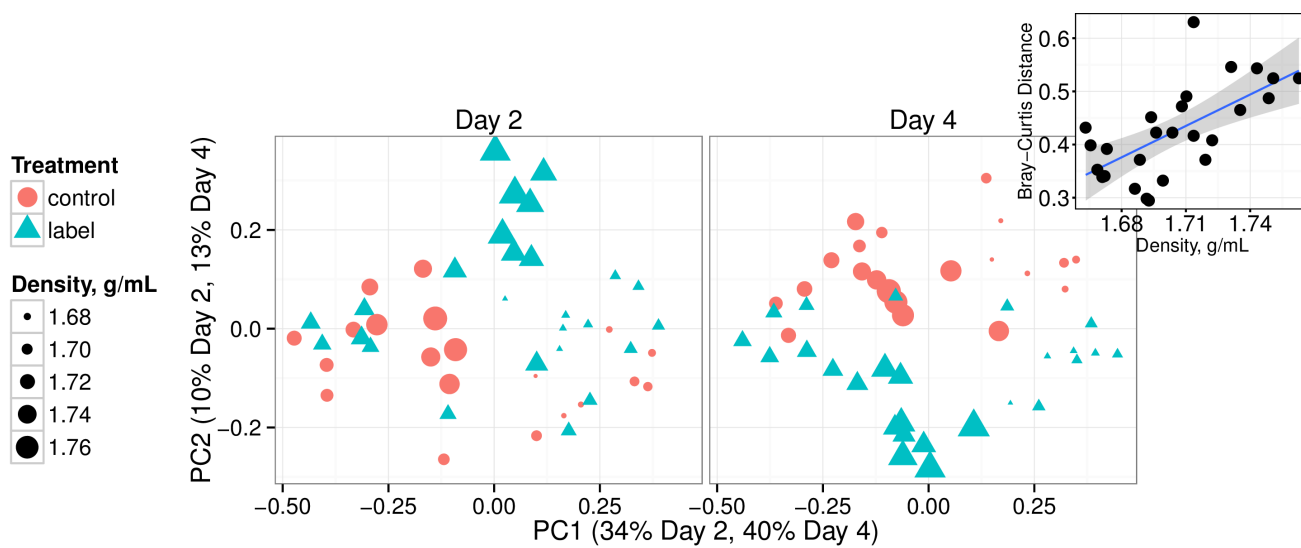
Figure S2. Ordination of Bray-Curtis sample pairwise distances for each incubation time. Point area is proportional to the density of the CsCl gradient fraction for each sequence library, and color/shape reflects control (red triangles) or labeled (blue circles) treatment. Inset shows Bray-Curtis distances for paired control versus labeled CsCl gradient fractions (i.e. fractions from the same incubation day and same density) against the density of the pair (p-value: $4.526e^{-5}$, r^2: 0.434).
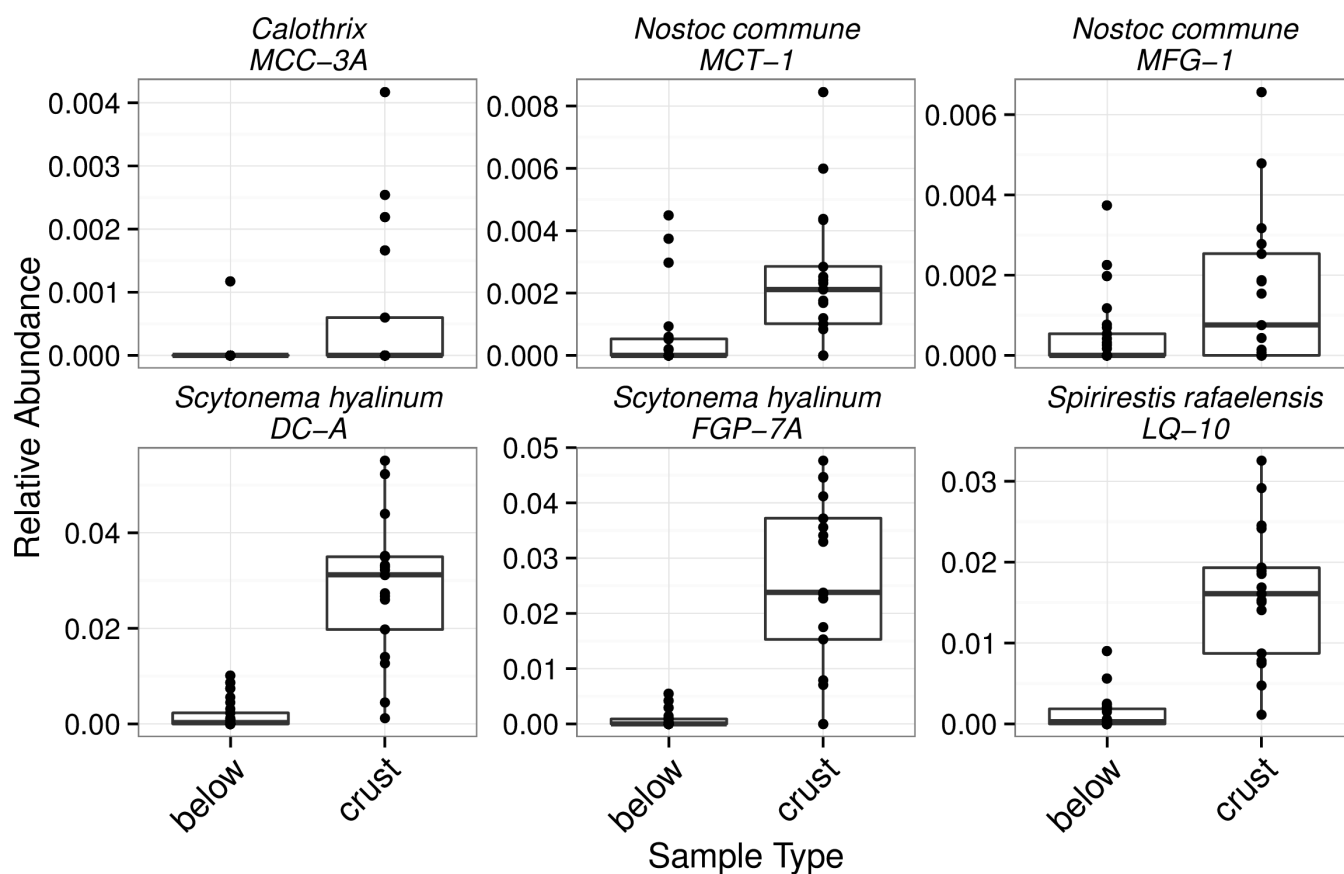
Figure S3. Relative abundance of selected heterocystous cyanobacterial OTUs with centroids from sequences described in Yeager et al. (2006) (see methods for selection criteria) in Steven et al. (2013) data set.
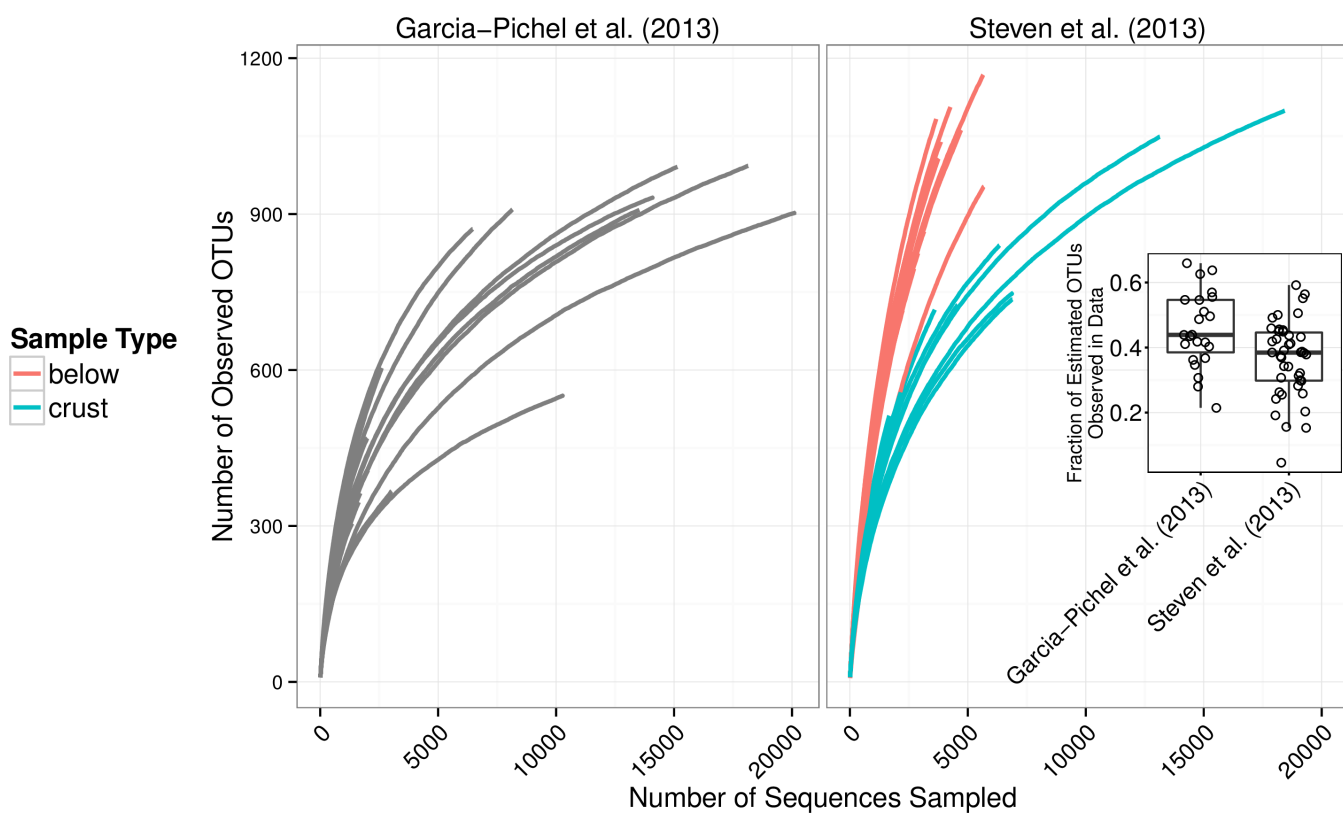
Figure S4. Rarefaction curves for all samples presented by Garcia-Pichel et al. (2013) and Steven et al. (2013). Inset is boxplot of estimated sampling effort for all samples in Garcia-Pichel et al. (2013) and Steven et al. (2013) (number of observed OTUs divided by number of CatchAll Bunge (2010) estimated total OTUs)
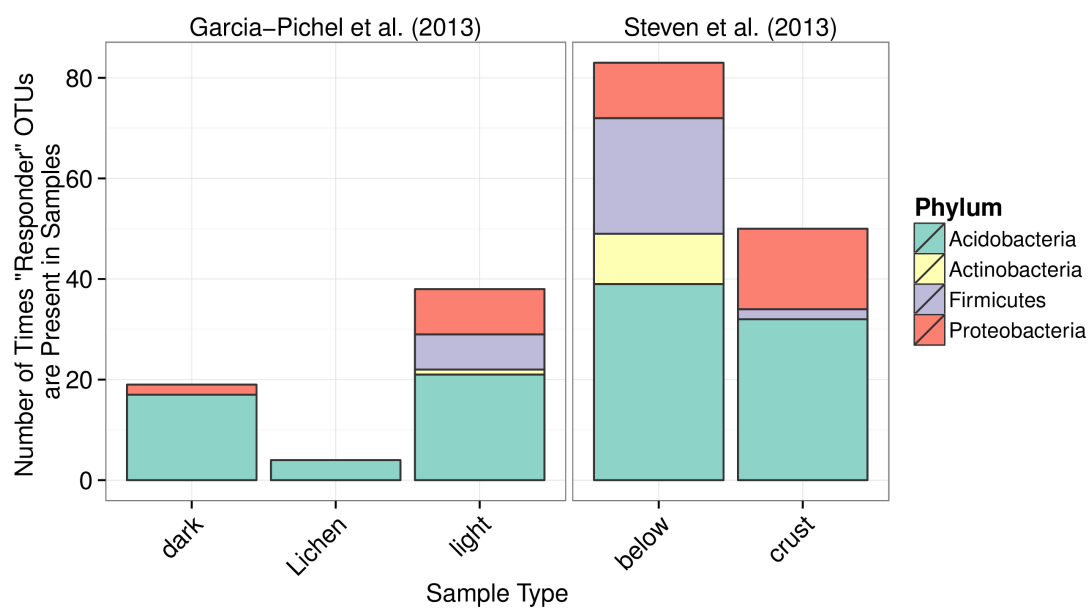
Figure S5. Counts of "responder" OTU occurrences in samples from Steven et al. (2013) and Garcia-Pichel et al. (2013). Steven et al. (2013) collected BSC samples (25 samples total) and samples from soil beneath BSC (17 samples total, "below" column in figure). Garcia-Pichel et al. (2013) collected samples from "dark" (9 samples total) and "light" (12 samples total) crusts in addition to "lichen" (2 samples total) dominated crusts.

Table 1.^{15}N responders BLAST against Living Tree Project

OTU ID	*Species Name*	**BLAST percent identity**	**accession**
OTU.108	*Caloramator proteoclasticus*	96.94	X90488
OTU.14	*Pantoea rwandensis*	99.49	JF295055
	Pantoea rodasii	99.49	JF295053
	Kluyvera intermedia	99.49	AF310217
	Kluyvera cryocrescens	99.49	AF310218
	Klebsiella variicola	99.49	AJ783916
	Klebsiella pneumoniae subsp. rhinoscleromatis	99.49	Y17657
	Klebsiella pneumoniae subsp. pneumoniae	99.49	X87276
	Erwinia aphidicola	99.49	FN547376
	Enterobacter soli	99.49	GU814270
	Enterobacter ludwigii	99.49	AJ853891
	Enterobacter kobei	99.49	AJ508301
	Enterobacter hormaechei	99.49	AJ508302
	Enterobacter cloacae subsp. dissolvens	99.49	Z96079
	Enterobacter cancerogenus	99.49	Z96078
	Enterobacter asburiae	99.49	AB004744
	Enterobacter amnigenus	99.49	AB004749
	Enterobacter aerogenes	99.49	AB004750
	Buttiauxella warmboldiae	99.49	AJ233406
	Buttiauxella noackiae	99.49	AJ233405
	Buttiauxella izardii	99.49	AJ233404
	Buttiauxella agrestis	99.49	AJ233400
OTU.1673	*Clostridium drakei*	95.9	Y18813
	Clostridium carboxidivorans	95.9	FR733710
OTU.327	*Clostridium hydrogeniformans*	94.92	DQ196623
	Clostridium amylolyticum	94.92	EU037903
OTU.330	*Clostridium lundense*	96.94	AY858804
OTU.342	*Acinetobacter johnsonii*	100.0	Z93440
OTU.4037	*Fonticella tunisiensis*	93.85	HE604099
OTU.54	*Shigella sonnei*	100.0	FR870445
	Shigella flexneri	100.0	X96963
	Escherichia fergusonii	100.0	AF530475
	Escherichia coli	100.0	X80725
OTU.57	*Fonticella tunisiensis*	93.88	HE604099
	Caloramator proteoclasticus	93.88	X90488
OTU.586	*Vitreoscilla filiformis*	98.48	HM037993
	Ottowia pentelensis	98.48	EU518930
	Ideonella dechloratans	98.48	X72724
	Diaphorobacter nitroreducens	98.48	AB064317
	Comamonas terrigena	98.48	AF078772