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Linking soil microbial community structure to potential carbon mineralization: A continental scale assessment of reduced tillage

Elizabeth L. Rieke
Soil Health Institute

Shannon B. Cappellazzi
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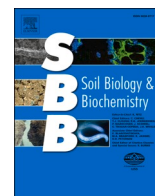
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Recommended Citation

Rieke, E.L., Cappellazzi, S.B., Cope, M., Liptzin, D., Greub, K.L.H., Norris, P.W., Tracy, E., Aberle, E., Bary, R.L., Baumhardt, A.B., Gracia, D.C., Brainard, J.R., Brennan, D.B., Reyes, D., Bruhjell, C.N., Carlyle, J.J.W., Crawford, C.F., Creech, S.W., Culman, B., Deen, C.J., Dell, J.D., Derner, T.F., Ducey, S.W., Duiker, M.F., Dyck, B.H., Ellert, A.E., Solorio, S.J., Fonte, S., Fonteyne, A.M., Fortuna, J.L., Foster, L.M., Fultz, S.E., Gampel, G.M., Godden, D., Griffin, LaHue, J.H., Grove, S.K., Hamilton, X., Hao, Z.D., Hayden, N., Honsdorf, J.A., Howe, J.A., Ippolito, G.A., Johnson, M.A., Kautz, N.R., Kitchen, S., Kumar, K.S.M., Kurtz, F.J., Larney, K.L., Lewis, M., Liebman, A.L., Ramirez, S., Machado, B., Maharjan, M.A., Martinez Gamiño, W.E., May, M.P., McClaran, M.D., McDaniel, N., Millar, J.P., Mitchell, A.D., Moore, P.A., Moore, M.M., Gutiérrez, K.A., Nelson, E.C., Omondi, S.L., Osborne, L.O., Alcalá, P., Owens, E.M., Pena-Yewtukhiw, H.J., Poffenbarger, B.P., Lira, J.R., Reeve, T.M., Reinbott, M.S., Reiter, E.L., Ritchey, K.L., Roozeboom, Y., Rui, A., Sadeghpour, U.M., Sainju, G.R., Sanford, W.F., Schillinger, R.R., Schindelbeck, M.E., Schipanski, A.J., Schlegel, K.M., Scow, L.A., Sherrod, A.L., Shober, S.S., Sidhu, E.S., Moya, M., St Luce, J.S., Strock, A.E., Suyker, V.R., Sykes, H., Tao, A.T., Campos, L.L., Van Eerd, N., Verhulst, T.J., Vyn, Y., Wang, D.B., Watts, B.B., William, D.L., Wright, T., Zhang, C.L.S., Morgan, C.W. Honeycutt, "Linking soil microbial community structure to potential carbon mineralization: A continental scale assessment of reduced tillage", *Soil Biology and Biochemistry*, Volume 168, 2022, 108618, ISSN 0038-0717, <https://doi.org/10.1016/j.soilbio.2022.108618>.

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Authors

Elizabeth L. Rieke, Shannon B. Cappellazzi, Michael Cope, Daniel Liptzin, Gregory Mac Bean, Kelsey L.H. Greub, Charlotte E. Norris, Paul W. Tracy, Ezra Aberle, Amanda Ashworth, Oscar Bañuelos Tavarez, Andy I. Bary, Roland L. Baumhardt, Alberto Borbón Gracia, Daniel C. Brainard, Jameson R. Brennan, Dolores Briones Reyes, Darren Bruhjell, Cameron N. Carlyle, James J.W. Crawford, Cody F. Creech, Steve W. Culman, Bill Deen, Curtis J. Dell, Justin D. Derner, Thomas F. Ducey, Sjoerd W. Duiker, Miles F. Dyck, Benjamin H. Ellert, Avelino Espinosa Solorio, Steven J. Fonte, Simon Fonteyne, Ann-Marie Fortuna, Jamie L. Foster, Lisa M. Fultz, Audrey V. Gamble, Charles M. Geddes, Deirdre Griffin-LaHue, John H. Grove, Stephen K. Hamilton, Xiyang Hao, Zachary D. Hayden, Nora Honsdorf, Julie A. Howe, James A. Ippolito, Gregg A. Johnson, Mark A. Kautz, Newell R. Kitchen, Sandeep Kumar, Kirsten S.M. Kurtz, Francis J. Larney, Katie L. Lewis, Matt Liebman, Antonio Lopez Ramirez, Stephen Machado, Bijesh Maharjan, Miguel Angel Martinez Gamiño, William E. May, Mitchel P. McClaran, Marshall D. McDaniel, Neville Millar, Jeffrey P. Mitchell, Amber D. Moore, Philip A. Moore, Manuel Mora Gutiérrez, Kelly A. Nelson, Emmanuel C. Omondi, Shannon L. Osborne, Leodegario Osorio Alcalá, Philip Owens, Eugenia M. Pena-Yewtukhiw, Hanna J. Poffenbarger, Brenda Ponce Lira, Jennifer R. Reeve, Timothy M. Reinbott, Mark S. Reiter, Edwin L. Ritchey, Kraig L. Roozeboom, Yichao Rui, Amir Sadeghpour, Upendra M. Sainju, Gregg R. Sanford, William F. Schillinger, Robert R. Schindelbeck, Meagan E. Schipanski, Alan J. Schlegel, Kate M. Scow, Lucretia A. Sherrod, Amy L. Shober, Sudeep S. Sidhu, Ernesto Solís Moya, Mervin St Luce, Jeffrey S. Strock, Andrew E. Suyker, Virginia R. Sykes, Haiying Tao, Alberto Trujillo Campos, Laura L. Van Eerd, Nele Verhulst, Tony J. Vyn, Yutao Wang, Dexter B. Watts, Bryan B. William, David L. Wright, Tiequan Zhang, Cristine L.S. Morgan, and C. Wayne Honeycutt



Linking soil microbial community structure to potential carbon mineralization: A continental scale assessment of reduced tillage

Elizabeth L. Rieke^{a,*}, Shannon B. Cappellazzi^a, Michael Cope^a, Daniel Liptzin^a, G. Mac Bean^a, Kelsey L.H. Greub^a, Charlotte E. Norris^a, Paul W. Tracy^a, Ezra Aberle^b, Amanda Ashworth^c, Oscar Bañuelos Tavarez^d, Andy I. Bary^e, R.L. Baumhardt^f, Alberto Borbón Gracia^g, Daniel C. Brainard^h, Jameson R. Brennanⁱ, Dolores Briones Reyes^g, Darren Bruhjell^j, Cameron N. Carlyle^k, James J.W. Crawford^l, Cody F. Creech^m, Steve W. Culmanⁿ, Bill Deen^o, Curtis J. Dell^p, Justin D. Derner^q, Thomas F. Ducey^r, Sjoerd W. Duiker^s, Miles F. Dyck^t, Benjamin H. Ellert^u, Avelino Espinosa Solorio^v, Steven J. Fonte^w, Simon Fonteyne^d, Ann-Marie Fortuna^x, Jamie L. Foster^y, Lisa M. Fultz^z, Audrey V. Gamble^{aa}, Charles M. Geddes^u, Deirdre Griffin-LaHue^{ab}, John H. Grove^{ac}, Stephen K. Hamilton^{ad}, Xiyang Hao^u, Zachary D. Hayden^h, Nora Honsdorf^{ae}, Julie A. Howe^{af}, James A. Ippolito^w, Gregg A. Johnson^{ag}, Mark A. Kautz^{ah}, Newell R. Kitchen^{ai}, Sandeep Kumar^{aj}, Kirsten S. M. Kurtz^{ak}, Francis J. Larney^u, Katie L. Lewis^{al}, Matt Liebman^{am}, Antonio Lopez Ramirez^{an}, Stephen Machado^{ao}, Bijesh Maharjan^m, Miguel Angel Martinez Gamiño^g, William E. May^{ap}, Mitchel P. McClaran^{aq}, Marshall D. McDaniel^{am}, Neville Millar^{ad}, Jeffrey P. Mitchell^{ar}, Amber D. Moore^{as}, Philip A. Moore Jr.^c, Manuel Mora Gutiérrez^g, Kelly A. Nelson^{at}, Emmanuel C. Omondi^{au}, Shannon L. Osborne^{av}, Leodegario Osorio Alcalá^g, Philip Owens^c, Eugenia M. Pena-Yewtukhiw^{aw}, Hanna J. Poffenbarger^{ax}, Brenda Ponce Lira^{ay}, Jennifer R. Reeve^{az}, Timothy M. Reinbott^{ba}, Mark S. Reiter^{bb}, Edwin L. Ritchey^{ac}, Kraig L. Roozeboom^{bc}, Yichao Rui^{bd}, Amir Sadeghpour^{be}, Upendra M. Sainju^{bf}, Gregg R. Sanford^{bg}, William F. Schillinger^{bh}, Robert R. Schindelbeck^{ak}, Meagan E. Schipanski^w, Alan J. Schlegel^{bi}, Kate M. Scow^{bj}, Lucretia A. Sherrod^{bk}, Amy L. Shober^{bl}, Sudeep S. Sidhu^{bm}, Ernesto Solís Moya^g, Mervin St Luce^{bn}, Jeffrey S. Strock^{bo}, Andrew E. Suyker^{bp}, Virginia R. Sykes^{bq}, Haiying Tao^{br}, Alberto Trujillo Campos^g, Laura L. Van Eerd^{bs}, Nele Verhulst^d, Tony J. Vyn^{bt}, Yutao Wang^{bu}, Dexter B. Watts^{bv}, Bryan B. Williams^{aw}, David L. Wright^{bm}, Tiequan Zhang^{bu}, Cristine L.S. Morgan^a, C. Wayne Honeycutt^a

^a Soil Health Institute, 2803 Slater Road, Suite 115, Morrisville, NC, 27560, USA

^b North Dakota State University Carrington Research Extension Center, 663 Highway 281 NE, Carrington, ND, 58421, USA

^c USDA ARS Poultry Production and Product Safety Research Unit, O-303 Poultry Science Center, Fayetteville, AR, 72701, USA

^d International Maize and Wheat Improvement Center (CIMMYT) Carretera México-Veracruz, km 45, Texcoco, MEX, 56237, Mexico

^e Washington State University Department of Crop and Soil Sciences, 2606 W Pioneer Avenue, Puyallup, WA, 98371, USA

^f USDA ARS Soil and Water Management Research Unit, 2300 Experiment Station Road, Bushland, TX, 79012, USA

^g Instituto Nacional de Investigaciones Forestales, Agrícolas y Pecuarias (INIFAP) Progreso 5, Santa Catarina Coyoacán, CMX, 04010, Mexico

^h Michigan State University Department of Horticulture, 1066 Bogue Street, East Lansing, MI, 48824, USA

ⁱ South Dakota State University West River Ag Center, 711N Creek Drive, Rapid City, SD, 57703, USA

^j Agriculture and Agri-Food Canada Edmonton, AB, T5J 4C3, Canada

Abbreviations: ASV, amplicon sequence variant; Cmin, 24-h potential carbon mineralization; PERMANOVA, permutational multivariate analysis of variance; NAPESHM, North American Project to Evaluate Soil Health Measurements.

* Corresponding author.

E-mail address: erieke@soilhealthinstitute.org (E.L. Rieke).

<https://doi.org/10.1016/j.soilbio.2022.108618>

Received 13 September 2021; Received in revised form 31 January 2022; Accepted 8 February 2022

Available online 11 March 2022

0038-0717/© 2022 The Authors.

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- ^k University of Alberta Department of Agriculture, Food and Nutritional Science Edmonton, AB, T6G 2P5, Canada
- ^l University of Missouri Extension Agricultural Engineering, 201 Highway 136, East Rock Port, MO, 64482, USA
- ^m University of Nebraska-Lincoln Panhandle Research and Extension Center, 4502 Avenue I, Scottsbluff, NE, 69361, USA
- ⁿ The Ohio State University School of Environment & Natural Resources, 130 Williams Hall, 1680 Madison Avenue, Wooster, OH, 44691, USA
- ^o University of Guelph Department of Plant Agriculture Guelph, ON, N1G 2W1, Canada
- ^p USDA ARS Pasture Systems & Watershed Management Research Unit, Building 3702, Curtin Road University Park, PA, 16802, USA
- ^q USDA ARS Rangeland Resources and Systems Research Unit, 8408 Hildreth Road, Cheyenne, WY, 82009, USA
- ^r USDA ARS Coastal Plains Soil, Water, & Plant Research Center, 2611 W Lucas Street, Florence, SC, 29501, USA
- ^s Pennsylvania State University Department of Plant Science, 408 Agricultural Sciences and Industries Building University Park, PA, 16802, USA
- ^t University of Alberta Department of Renewable Resources Edmonton, AB, T6G 2H1, Canada
- ^u Agriculture and Agri-Food Canada Lethbridge Research and Development Centre, Lethbridge, AB, T1J 4B1, Canada
- ^v Sustentabilidad Agropecuaria Querétaro, SAQ Calle Motolinia 17, Cimatario Santiago de Querétaro, QUE, 76030, Mexico
- ^w Colorado State University Department of Soil and Crop Sciences, 307 University Avenue, Fort Collins, CO 80523, USA
- ^x USDA ARS Grazinglands Research Lab, 7207 W Cheyenne Street, El Reno, OK, 73036, USA
- ^y Texas A&M AgriLife Research, 3507 Hwy 59 E, Beeville, TX, 78102, USA
- ^z Louisiana State University School of Plant, Environment & Soil Sciences, 104 M.B. Sturgis Hall Baton Rouge, LA, 70803, USA
- ^{aa} Auburn University Department of Crop, Soil and Environmental Sciences, 202 Funchess Hall Auburn University, AL, 36849, USA
- ^{ab} Washington State University Department of Crop and Soil Sciences, 16650 WA-536, Mount Vernon, WA, 98273, USA
- ^{ac} University of Kentucky Department of Plant and Soil Science, Research and Education Center, 1205 Hopkinsville Street, Princeton, KY, 42445, USA
- ^{ad} Michigan State University W.K. Kellogg Biological Station, 3700 E Gull Lake Drive Hickory, Corners, MI, 49060, USA
- ^{ae} Kiel University Institute of Crop Science and Plant Breeding Christian-Albrechts-Platz 4 Kiel, 24118, Germany
- ^{af} Texas A&M AgriLife Research Department of Soil and Crop Sciences MS, 2474, Heep Center, 370 Olsen Boulevard, College Station, TX, 77843, USA
- ^{ag} University of Minnesota Southern Research and Outreach Center, 35838 120th Street, Waseca, MN, 56093, USA
- ^{ah} USDA ARS Southwest Watershed Research Center, 2000 E Allen Road, Tucson, AZ, 85719, USA
- ^{ai} USDA ARS USDA-ARS Cropping Systems and Water Quality Research Unit, 243 Agricultural Engineering Building, Columbia, MO, 65211, USA
- ^{aj} South Dakota State University Department of Agronomy, Horticulture and Plant Science, Room 248C NPB, BOX 2140C, 1110 Rotunda Lane North, Brookings, SD, 57007, USA
- ^{ak} Cornell University Department of Soil and Crop Sciences, 306 Tower Road, Ithaca, NY, 14853, USA
- ^{al} Texas A&M AgriLife Research Department of Soil and Crop Sciences, 1102 East FM 1294, Lubbock, TX, 79403, USA
- ^{am} Iowa State University Department of Agronomy, 716 Farm House Lane, Ames, IA, 50011, USA
- ^{an} Centro de Bachillerato Tecnológico Agropecuario No. 305, CBTA 305 Prolongación 9 Sur, Molcaxac, PUE, 75650, Mexico
- ^{ao} Oregon State University Columbia Basin Agricultural Research Center, 48037 Tubbs Ranch Road, Adams, OR, 97810, USA
- ^{ap} Agriculture and Agri-Food Canada Canada, Indian Head Research Farm Indian Head, SK, S0G 2K0, Canada
- ^{aq} University of Arizona School of Natural Resources & the Environment Tucson, AZ, 85721, USA
- ^{ar} University of California, Davis Department of Plant Sciences, 9240 S. Riverbend Avenue, Parlier, CA, 93648, USA
- ^{as} Oregon State University Department of Crop and Soil Science, 2750 Campus Way, Corvallis, OR, 97331, USA
- ^{at} University of Missouri Division of Plant Sciences, 64399 Greenley Place, Novelty, MO, 63460, USA
- ^{au} Tennessee State University Department of Agricultural and Environmental Sciences Nashville, TN, 37209, USA
- ^{av} USDA ARS North Central Agricultural Research Laboratory, 2923 Medary Avenue, Brookings, SD, 57006, USA
- ^{aw} West Virginia University Davis College of Agriculture, Natural Resources and Design, 1194 Evansdale Drive, Morgantown, WV, 26506, USA
- ^{ax} University of Kentucky Department of Plant and Soil Science, 405 Veterans Drive, Lexington, KY, 40546, USA
- ^{ay} Universidad Politécnica de Francisco I, Madero (UPFIM) Ingeniería Agrotecnología Carretera Tepatepec-San Juan Tepa, km 2, Tepatepec, HID, 42660, Mexico
- ^{az} Utah State University Plants, Soils and Climate Department, 4820 Old Main Hill AGRS 328, Logan, UT, 84322, USA
- ^{ba} University of Missouri Bradford Research Center College of Agriculture, Food, and Natural Resources, 3600 E New Haven Road, Columbia, MO, 65201, USA
- ^{bb} Virginia Tech Eastern Shore Agricultural Research and Extension Center, 33446 Research Drive, Painter, VA, 23420, USA
- ^{bc} Kansas State University Department of Agronomy, 2004 Throckmorton Plant Sciences Center, 1712 Claffin Road, Manhattan, KS, 66506, USA
- ^{bd} Rodale Institute, 611 Siegfriedale Road, Kutztown, PA, 19530, USA
- ^{be} Southern Illinois University Crops, Soils, and Environmental Management Program, School of Agricultural Sciences, 1205 Lincoln Drive, Carbondale, IL, 62901, USA
- ^{bf} USDA ARS Northern Plains Agricultural Research Laboratory, 1500 North Central Avenue, Sidney, MT, 59270, USA
- ^{bg} University of Wisconsin Department of Agronomy, 1575 Linden Dr. Madison, WI, 53706, USA
- ^{bh} Washington State University Dryland Research Station, PO Box B, Lind, WA, 99341, USA
- ^{bi} Kansas State University SW Research, Extension Center 1474 State Highway 96, Tribune, KS, 67879, USA
- ^{bj} University of California, Davis Department of Land, Air, and Water Resources, One Shields Avenue, Davis, CA, 95616, USA
- ^{bk} USDA ARS Center for Agricultural Resources Research, 2150 Center Avenue, Building D, Suite 200, Fort Collins, CO, 80526, USA
- ^{bl} University of Delaware Plant and Soil Sciences, 531 South College Avenue, Newark, DE, 19716, USA
- ^{bm} University of Florida North Florida Research and Education Center, 155 Research Road, Quincy, FL, 32351, USA
- ^{bn} Agriculture and Agri-Food Canada Swift Current Research and Development Centre, 1 Airport Road Swift Current, SK, S9H 3X2, Canada
- ^{bo} University of Minnesota Department of Soil, Water and Climate, SW Research & Outreach Center, 23669 130th Street, Lambertson, MN, 56152, USA
- ^{bp} University of Nebraska-Lincoln School of Natural Resources, 807 South Hardin Hall, 3310 Holdrege Street, Lincoln, NE, 68583, USA
- ^{bq} University of Tennessee Department of Plant Sciences, 2505 E J Chapman Drive, Knoxville, TN, 37996, USA
- ^{br} Washington State University Department of Crop and Soil Sciences, 245 Johnson Hall, Pullman, WA, 99164, USA
- ^{bs} University of Guelph School of Environmental Sciences, Ridgeway Campus 120 Main St E, Ridgeway, ON, N0P 2C0, Canada
- ^{bt} Purdue University Agronomy Department, 915 W State Street, West Lafayette, IN, 47907, USA
- ^{bu} Agriculture and Agri-Food Canada Harrow Research and Development Center, 2585 Essex County Rd 20, Harrow, ON, N0R 1G0, Canada
- ^{bv} USDA ARS National Soil Dynamics Laboratory, Auburn, AL, 36832, USA

ARTICLE INFO

Keywords:

Microbial community
Potential carbon mineralization
Tillage
Soil health

ABSTRACT

Potential carbon mineralization (C_{min}) is a commonly used indicator of soil health, with greater C_{min} values interpreted as healthier soil. While C_{min} values are typically greater in agricultural soils managed with minimal physical disturbance, the mechanisms driving the increases remain poorly understood. This study assessed bacterial and archaeal community structure and potential microbial drivers of C_{min} in soils maintained under various degrees of physical disturbance. Potential carbon mineralization, 16S rRNA sequences, and soil characterization data were collected as part of the North American Project to Evaluate Soil Health Measurements (NAPESHM). Results showed that type of cropping system, intensity of physical disturbance, and soil pH influenced microbial sensitivity to physical disturbance. Furthermore, 28% of amplicon sequence variants (ASVs), which were important in modeling C_{min}, were enriched under soils managed with minimal physical

disturbance. Sequences identified as enriched under minimal disturbance and important for modeling C_{min}, were linked to organisms which could produce extracellular polymeric substances and contained metabolic strategies suited for tolerating environmental stressors. Understanding how physical disturbance shapes microbial communities across climates and inherent soil properties and drives changes in C_{min} provides the context necessary to evaluate management impacts on standardized measures of soil microbial activity.

1. Introduction

Over the past few decades numerous biologically based measurements have been designed to assess how reducing physical disturbance in row-cropping systems impact soil functioning (e.g., cycle nutrients, decompose organic matter) and overall soil health. Current measurements used by the scientific community to evaluate soil health include microbial biomass, available carbon and nitrogen pools for microbial consumption, and potential carbon and nitrogen mineralization (Acosta-Martinez et al., 2018; Culman et al., 2012; Gonzalez-Quiñones et al., 2011; Li et al., 2020; Norris et al., 2020). Greater values recorded in systems with reduced physical disturbance, when compared to intensively disturbed fields, are commonly interpreted under the assumption, “more is better” (Andrews et al., 2004; Haney et al., 2010, 2018; Moebius-Clune et al., 2016). However, greater values from these measurements are difficult to interpret because the measurements are not directly tied to increases in soil function, such as providing adequate plant nutrition or improved ecosystem health (e.g., increased carbon storage, reduced nitrogen losses) (Fierer et al., 2021). Understanding why these widely used measurements respond positively to adoption of reduced physical disturbance will allow for appropriate interpretations of the measurements and therefore allow stakeholders to understand how management choices affect soil function.

One measure of potential microbial activity related to soil health is potential carbon mineralization (C_{min}). Soil microbial community members respire carbon dioxide as a metabolic waste product while degrading organic matter and cycling nutrients. Standardized C_{min} assays report carbon dioxide fluxes following rewetting of air-dried, sieved soil under aerobic conditions (Zibilske, 2018), such as from either a 24- or 96-h incubation, among other modifications (Haney Soil Health Test or the Cornell Comprehensive Assessment of Soil Health, respectively) (Haney et al., 2010; Moebius-Clune et al., 2016). Potential carbon mineralization values are generally greater in systems employing reduced tillage across many soil types and climates (Nunes et al., 2020). However, the greater C_{min} measurements identified in the standardized laboratory assay conflict with *in situ* carbon mineralization measurements, where soils managed for reduced physical disturbance respire less carbon dioxide than their less disturbed counterparts (Abdalla et al., 2016). Nonetheless, C_{min} is often interpreted as *in situ* basal mineralization (Haney et al., 2018). Mineralization resulting from standardized C_{min} assays are a combination of the consumption of newly lysed cellular material, fresh metabolic material exuded during rewetting, and newly available organic residues following pretreatment of the soils (Fierer and Schimel, 2003; Kaiser et al., 2015). Identifying the groups of organisms responsible for driving greater C_{min} values in soils managed for minimal physical disturbance will help provide a scientifically backed interpretation of this already widely used measurement, rather than relying on the assumption, “more is better.”

While C_{min} generally increases in soils managed for minimal physical disturbance, the impact of physical disturbance on microbial community structure has varied among site-specific studies. Many studies of physical disturbance and community structure report significant changes in both community structure and community diversity in response to disturbance treatments (Ceja-Navarro et al., 2010; De Quadros et al., 2012; Schmidt et al., 2018; Sengupta and Dick, 2015; Srouf et al., 2020; Z. Wang et al., 2016), while just as many others report a significant change in only one of the two metrics (Navarro-Noya et al., 2013; Ng et al., 2012; Schlatter et al., 2019; Schmidt et al., 2019; Smith

et al., 2016; Yin et al., 2010). Such variability may be associated with differences in sampling time, tillage equipment, cropping system history, sample processing, statistical analyses, inherent soil properties, and climate factors. Understanding the impact of physical disturbance on soil microbial community structure across a range of climates, cropping systems, and inherent soil properties (e.g., texture, pH) may enhance interpretation of divergent results from site-specific studies.

Assessment of microbial community structure is capable of identifying changes in community composition due to agricultural management, but it does not provide context as to how the change affects soil functioning. Measuring change in microbial community structure and function, in unison, provides context as to whether changes in agricultural management alter microbial function as well as the potential drivers responsible for changes in function. To date, a number of studies have attempted to link management driven changes in soil microbial community composition to changes in carbon mineralization (Guo et al., 2019; Khanghahi et al., 2019; Liu et al., 2018; Malik et al., 2018; Mbuthia et al., 2015). These studies indicate that analyzing soil microbial community composition in addition to inherent soil properties and other biological measurements enhances the predictability of carbon mineralization. However, the goal of these studies was to uncover drivers of basal soil mineralization, which may differ from the organisms responsible for driving the burst of mineralization recorded in standardized C_{min} incubations. Uncovering the microbial community members responsible for driving greater C_{min} measurements in systems with reduced physical disturbance may provide much needed context to the measure, which in turn will allow stakeholders to appropriately synthesize their results in the context of building soil health.

The goal of the present study was to evaluate the impact of reduced physical disturbance on soil bacterial and archaeal community members and their potential influence on greater C_{min} measurements recorded in long-term reduced disturbance systems across major agricultural areas in North America. We hypothesized that bacterial and archaeal communities that were enriched under minimum physical disturbance would be important predictors of C_{min}. We first explored relationships between disturbance intensity and changes in bacterial and archaeal community structures. We identified a subset of ASVs which were enhanced in systems employing minimum disturbance management across a range of inherent soil properties and climates. Finally, we identified a suite of bacteria and archaeal taxa, which were enriched under minimum disturbance and important predictors of C_{min} models.

2. Methods

2.1. Sample collection

Data used in these analyses were collected as part of the North American Project to Evaluate Soil Health Measurements (NAPESHM). NAPESHM sites were chosen based on the presence of treatments to test the management effects of tillage, cover crops, crop rotation, nutrient amendments, irrigation, and livestock stocking rate and intensity. A full description of the project can be found in Norris et al. (2020). The project consisted of 2032 experimental units from 688 replicated treatments located at 124 long term experimental agricultural research sites across North America (Fig. 1). Out of the 688 replicated treatments, 568 treatments contained at least ten years of consistent crop rotations and agricultural management practices (e.g. tillage, cover cropping), while 120 treatments contained between six and ten years of consistent

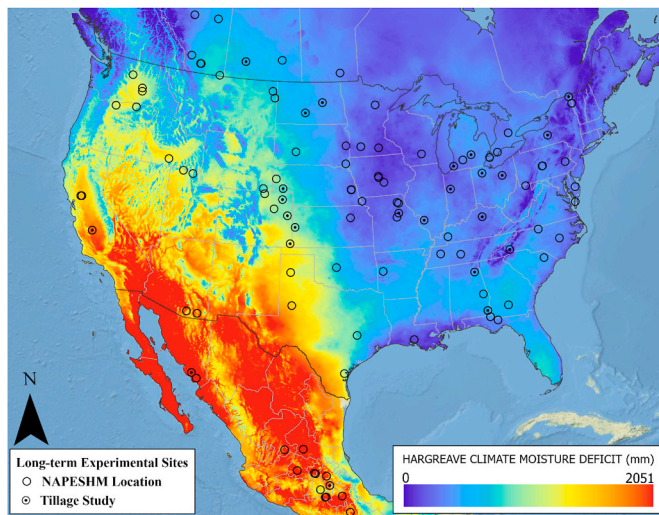


Fig. 1. Experimental unit sampling locations included in the North American Project to Evaluate Soil Health Measurements (NAPESHM). Concentric circles indicate sites containing at least 12 experimental units with paired tillage treatments. The map is colored by Hargreaves Climate Moisture Deficit.

practices. Detailed treatment and site information is located in [Table 1](#), [Supplemental Table 1](#), and in [Norris et al. \(2020\)](#).

Sites were predominantly sampled in spring of 2019 prior to fertilization, spring tillage, and planting. Five-year detailed management histories were collected for each treatment. Soil sampling for each experimental unit was performed using a sharpshooter shovel and soil knife. A total of 18 knife slices (15- by 4-cm) were collected uniformly to a depth of 15 cm from six locations across each experimental unit in a zig-zag pattern and placed on ice. Samples were composited in a bucket prior to bagging and shipping. Sampling equipment was cleaned and sterilized with isopropyl alcohol between experimental units. Additionally, sterile nitrile gloves were worn during sampling and sample processing.

2.2. Site climate data

The Hargreaves Climate Moisture Deficit was calculated to capture the combined effect of precipitation and temperature ([Hargreaves and Allen, 2003](#)). The moisture deficit calculations included a monthly estimate of precipitation deficit averaged from 1991 to 2020 ([T. Wang et al., 2016](#)). The monthly deficit for a given location represented the difference between reference evapotranspiration and precipitation, and the deficit is considered zero for any month where precipitation is greater than reference evapotranspiration.

2.3. Laboratory measurements

For each experimental unit, composite soil samples were sent to the Soil Water and Environmental Lab¹ at Ohio State University for measurement of particle size distribution, pH, total nitrogen, total carbon, inorganic carbon, and Cmin. Particle size analyses were performed using the pipette method and sands were wet sieved ([Gee and Or, 2018](#)). Soil pH measurements were made using a 1:2 soil:water slurry with a pH electrode ([Campbell et al., 2018](#)). Total nitrogen and carbon were measured by dry combustion ([Nelson and Sommers, 2015](#)). Inorganic carbon was measured using Chittick gasometric calcimeter^a (St. Louis, MO, USA). Soil organic carbon was calculated as the difference between

Table 1
Primary tillage implements used in disturbance category analyses.

Site ID	Disturbance Category			Crop Rotation
	Minimum	Moderate	Intense	
CASK01	Planting only	N/A	Cultivate	Spring wheat-fallow; wheat-pea, chickpea, lentil pulse
MXEM01	Planting only	N/A	Disk harrow, chisel plow	Continuous corn; continuous wheat; wheat-corn
MXSO03	Planting only	N/A	Disk harrow, chisel plow	Continuous spring wheat; wheat-corn
USAL01	Planting only	N/A	Rototill, disk	Corn-soybean
USAL03	N/A ^a	Strip-till	Moldboard	bahiagrass-bahiagrass-peanut-cotton; peanut-cotton
USCA03	Planting with fertilizer shank	N/A	Subsoiler, disk harrow, bedder	Garbanzo bean-sorghum
USIL02	Planting only	Chisel plow, disk harrow, row cultivate	Moldboard, disk harrow, cultivate	Corn-soybean
USIN01	Planting only	Chisel plow, disk harrow, row cultivate	Moldboard, disk harrow, cultivate	Continuous corn; continuous soybean; corn-soybean
USKY03	Planting only	N/A	Moldboard, disk harrow	Continuous corn
USMI01	N/A	Strip-till, row cultivate, disc harrow	Disk harrow, rototill, row cultivate	Specialty vegetables
USMI02	N/A	Spring strip-till, fall disc harrow	Moldboard, disk harrow	Specialty vegetables
USMO01	Planting only	Biennial spring chisel and fall disc harrow	N/A	Corn-soybean
USNC01	Planting only	N/A	Chisel plow and rotary hoe	Corn-soybean-sorghum
USND01	Planting only	Spring cultivate	N/A	Spring wheat-fallow; spring wheat-corn; spring wheat-corn-soybean
USND02	Planting only	Fall disk harrow and chisel plow; Spring cultivate every four year	Chisel plow, disk harrow, cultivate	Spring wheat-field pea- corn-soybean
USNY01	Planting with double disk fertilizer opening	N/A	Moldboard, cultivate	Continuous corn
USNY04	Planting with double disk fertilizer opening	N/A	Moldboard, cultivate	Continuous corn
USOH02	Planting only	Chisel and rototill	Moldboard, rototill	Corn; corn-soybean; corn-alfalfa-alfalfa
USOH03	Planting only	Chisel and cultivate	Moldboard, disk harrow, cultivate	Corn; corn-soybean; corn-red clover-red clover

^a Treatment was not present at study location.

¹ Name is given to provide specific information and does not constitute endorsement by the authors over other entities that may be equally suitable.

total carbon and inorganic carbon (Dreimanis, 1962). Soil samples used in Cmin measurements were air dried and passed through a 2-mm sieve prior to incubation. Potential carbon mineralization was measured as accumulation of CO₂-C following rewetting by capillary action and a 24-h incubation period (Zibilske, 2018).

2.4. DNA extraction and sequencing

Composite moist soil samples from each experimental unit were passed through a sterile 8-mm sieve and shipped overnight on ice to the Center for Genome Research and Biocomputing at Oregon State University for DNA extraction and subsequent sequencing. From each experimental unit, 0.25 g of soil was weighed for DNA extraction. DNA extractions were performed using a Thermo KingFisher Flex robotic magnetic bead system^a (Swindon, UK) with Qiagen MagAttract Power Soil DNA kits^a (Germantown, MD, USA). A Zymobiotics microbial community standard was included on each DNA extraction plate. DNA concentrations were quantified fluorometrically using the Invitrogen Quant-iT dsDNA High Sensitivity Assay Kit^a (Waltham, Massachusetts, USA) and read on a BioTek Synergy HT microplate reader^a (Winooski, VT, USA).

Following Earth Microbiome Project protocols (Caporaso et al., 2018), the V4 region of 16S rRNA was amplified and sequenced using the Illumina MiSeq platform^a. The QIIME 2 platform was used for classification of reads to taxa counts (v. 2019.4). Adapters were trimmed with the qiime cutadapt trim-paired tool. Read pairs were merged with qiime vsearch join-pairs with a maximum merge length of 256 base pairs and removing any reads with any Ns present. Reads were filtered for a minimum PHRED score of 20. QIIME deblur denoise-16S was used to denoise the reads by removing reads that did not have a sequence similarity of 60% to the 85% OTU GreenGenes database and reads were trimmed to 250 base pairs or discarded if shorter. Results were tabulated with the qiime feature-table tabulate-seqs. Taxonomy were assigned with the QIIME feature-classifier classify-sklearn using the silva database classifier version 132. Data was exported from QIIME2 with QIIME tools export. Amplicon sequence variants that were observed less than three times in 5% of samples were removed. Remaining samples were rarified to 8000 sequences using the function rarify_even_depth in the R package phyloseq (McMurdie and Holmes, 2013). Rarified samples were used in all downstream analyses. Demultiplexed sequences were deposited in the National Center for Biotechnology Information Sequence Read Archive with the following accession number: PRJNA762046.

2.5. Statistical analysis

In addition to treatments that represented management changes, sites included in the study cover a broad range of climates, inherent soil properties, and organic matter. To identify significant differences in community structure as a result of differences in inherent soil properties and climate, permutational multivariate analysis of variance (PERMANOVA) with distance matrices was performed using the Adonis function in the R package vegan (Dixon, 2003). Canonical correspondence analysis was also performed to visualize how inherent soil properties and climate were related to community structure using the ordinate function. Alpha diversity was assessed by calculating Shannon Diversity Index values for all experimental units. Shannon Diversity and richness were calculated using the estimate_richness function in the R package phyloseq (McMurdie and Holmes, 2013). Experimental units with extreme Shannon Diversity and richness outliers were removed from the dataset.

A subset of replicated, paired tillage treatments was selected from nineteen sites for specific disturbance-based analysis (Fig. 1, Table 1). For this analysis, only sites which contained at least twelve experimental units evenly distributed across two or more tillage regimes were included in disturbance analyses. Furthermore, treatments were only included in disturbance analyses if a paired treatment existed, that is the

only difference in management was physical disturbance through tillage. Treatments were classified into minimum, moderate, or intense disturbance categories. Minimum disturbance included experimental units whose only physical soil disturbance occurred during planting, commonly referred to as “no-till”. Moderate disturbance encompassed a wide range of reduced or conservation tillage practices, including strip tillage, row cultivation, and chisel plow. Intense disturbance included experimental units from treatments commonly described as “conventional tillage” where practices are among the most disruptive tillage practices for a given cropping system and climate. Sites included in tillage analyses were assigned a six-digit unique identifier (Supplementary Table 1). Additionally, treatments that only differed by tillage were assigned a two-digit unique identifier to ensure direct comparison of the treatments in downstream analyses. To visualize differences in community structure among disturbance categories, detrended correspondence analyses (DCAs) were performed with bray-curtis distance matrices to account for non-linear relationships. Significant differences in community structure as a function of physical disturbance at individual sites were assessed using adonis function in the vegan package (Dixon, 2003). If a site contained a factorial design (e.g. tillage and cover cropping), the non-tillage factor was accounted for using the strata argument to ensure the direct comparison of treatments that only differed by tillage regimes. Permutational analysis of multivariate dispersions were assessed prior to implementing PERMANOVA using the betadisp function in the vegan package (Dixon, 2003). Non-significant values ($p > 0.05$) confirmed that the paired treatments contained similar within treatment variances, indicating significant differences in beta diversity were due to differences in physical disturbance treatments.

Experimental units from sites with significant differences in community structure due to physical disturbance, were used to identify ASVs whose abundances were significantly enriched in reduced disturbance treatments across all sites. Enrichment of specific ASVs was determined by differential expression analyses based on the negative binomial distribution (Gamma-Poisson), performed using the function Deseq from the R package DESeq2 (Love et al., 2018) with Wald tests as the test argument. Amplicon sequence variants were considered differentially expressed when $p < 0.05$. Three separate analyses were performed to identify: 1) ASVs that were enriched in minimum disturbance treatments in relation to intense disturbance treatments, 2) ASVs that were enriched in minimum disturbance treatments in relation to moderate disturbance treatments, and 3) ASVs that were enriched in moderate disturbance treatments in relation to intense disturbance treatments. The analysis was performed on rarefied experimental units due to large differences among experimental unit library sizes (~10x) (Weiss et al., 2017). Lastly, differences in alpha diversity were assessed separately between the three disturbance comparisons described above using the lm function contained within the base R package. In each analysis, disturbance category was set as a fixed effect and tillage treatment comparison as random effect using the assigned two-digit unique identifiers, therefore permitting comparison of treatments which only differed by physical disturbance. Model outputs were assessed using the anova function, contained within the base R package, to identify significant differences between disturbance categories.

Random forest regression models were employed to identify microbial drivers of Cmin measurements. Thirty random forest regressor models were fit to the full set of 689 experimental treatments to help sort and filter ASVs that were most highly associated with Cmin. In each model run, Cmin was the response variable, with ASVs as predictor variables. The ASVs included in the final set models were pruned by average abundance until total model permutation importance began to decline, resulting in 328 ASVs contained in each model. Training and testing datasets were built at random for each model iteration, with experimental units from 586 to 608 treatments randomly selected for inclusion in the training dataset, resulting experimental units from 80 to 102 treatments placed in the testing dataset. The set of fitted models had

an average R^2 of 0.58 predicting on testing data sets, which were held out from model training. The average ASV importance across models was used to estimate the relative utility of each ASV for predicting on testing data sets. Here, ASV importance is the difference between the initial prediction fit and the prediction fit after randomly permuting the respective ASV data. These analyses were performed in the Scikit-learn Python module ((Pedregosa et al., 2011); version 0.23.1, <https://scikit-learn.org/stable/>). The maximum number of ASVs allowed in any regression tree was 18 ($328^{1/2}$), with minimization of the mean squared residual error as the model criteria. The relationships between individual ASVs contained in the top decile of importance and Cmin were explored using linear regression. Code for analyses is located at <https://github.com/erieke/NAPESHM-tillage-mineralization>.

3. Results

3.1. Continental community and cmin assessment

Experimental units covered a wide range of inherent soil properties and climates (Table 2). Following rarefaction, 1924 experimental units remained in the dataset, consisting of 5322 unique ASVs. Twenty-seven phyla were identified in the dataset, with major contributions from *Acidobacteria*, *Actinobacteria*, *Bacteroidetes*, *Proteobacteria*, and *Verrococcinobacteria*. Shannon's diversity index ranged from 4.36 to 6.70, while observed ASV richness ranged from 342 to 1580. Greatest diversity and richness were observed at moderate pH values (5.5–7.5) and humid climates (Fig. 2). The PERMANOVA among communities across the continent indicated pH and climate moisture deficit explained more variation than clay, sand, soil organic carbon and total nitrogen (Table 2). Furthermore, the canonical correspondence analysis results suggest that climate and inherent soil properties predict only a small fraction of microbial community structure (Supplementary Fig. 1).

An in-depth analysis of Cmin in relation to climate, inherent soil properties, carbon-based soil health indicators, and agricultural management can be found in Liptzin et al. (Liptzin et al., n.d.). Briefly, measures of Cmin ranged from 4.7 to 126.7 mg C kg⁻¹d⁻¹, with a median of 49.0 mg C kg⁻¹d⁻¹. Potential carbon mineralization and soil organic carbon were moderately related ($r = 0.58$). Prediction of Cmin with inherent soil properties and climatic variables using multiple linear regression resulted in a R^2 value of 0.27. Clay, pH and precipitation were positively correlated with the measurement, while temperature was negatively correlated. Response ratios of paired treatments indicated significantly greater Cmin in systems employing reduced tillage, cover cropping, application of organic nutrients, or residue retention.

3.2. Microbial response to tillage intensity

Out of the 14 sites that contained at least 12 experimental units with minimum and intense disturbance comparisons, 11 contained significantly different microbial community structures ($p < 0.01$) as a result of differences in disturbance, which were identified using the adonis function (Fig. 3, Supplementary Table 2). The three sites with non-significant differences ($p > 0.01$) all contained wheat-centric rotations

Table 2

16S rRNA permutational multivariate analysis of laboratory soil measurements and climate. All measurement analyses were significant ($p < 0.01$).

Measurement	r^2	n^a	Minimum	Maximum	Median
Clay (%)	0.04	1909	2	63	21
Sand (%)	0.01	1909	3	91	32
pH	0.12	1909	4.0	9.0	6.3
Soil Organic Carbon (%)	0.02	1909	0.20	10.42	1.49
Hargreave Climate	0.07	1909	93	1773	259
Moisture Deficit (mm)					
Total Nitrogen (%)	0.02	1903	0.02	1.07	0.14

^a Number of samples included in the analysis.

(Table 1). Differential abundance testing of the 214 experimental units from the 11 sites identified 717 ASVs whose abundances were significantly greater in minimum disturbance experimental units ($p < 0.05$). The associated ASVs represent 119 of 242 families identified in both minimum and intense disturbance experimental units. On average, the ASVs accounted for 16% of sequences associated with intense disturbance experimental units, and 33% of sequences were associated with minimum disturbance. The 717 ASVs were confined within 14 phyla, with most sequences associated with *Acidobacteria*, *Actinobacteria*, *Bacteroidetes*, *Gemmatimonadetes*, *Proteobacteria*, *Rokubacteria*, and *Verrococcinobacteria* (Fig. 4). Alpha diversity, represented by the Shannon Diversity Index, and richness were not significantly different ($p > 0.10$) between minimum and intense disturbance categories (Supplementary Fig. 2).

While most minimum and intense disturbance comparisons returned significant differences in community structure at the site level, comparisons between minimum and moderate disturbance returned mixed results. Among the minimum and moderate disturbance comparisons, four of seven sites had significant differences ($p < 0.01$) in bacterial and archaeal community structures which were identified using the adonis function (Supplementary Table 3). The four sites with significant differences in community structure all contained a site average pH ≤ 5.7 or pH > 6.5 (Supplementary Table 3). Differential abundance testing of the 76 experimental units from the four sites returned 242 ASVs whose abundances were enriched ($p < 0.05$) in minimum compared to moderate disturbance. In general, the resulting ASVs accounted for 14% of ASV relative abundance in minimum disturbance and 4% of ASV relative abundance in moderate disturbance. Of the 75 bacterial and archaeal families enriched under minimum disturbance compared to moderate disturbance, 73 were also enriched in minimum compared to intense disturbance. Lastly, alpha diversity and richness were not significantly different ($p > 0.10$) between minimum and moderate disturbance (Supplementary Fig. 3).

Sites with moderate and intense physical disturbance comparisons returned a similar percentage of site level differences to minimum-moderate disturbance comparisons, with four of eight sites containing significant differences due to disturbance ($p < 0.01$) (Supplementary Table 4) identified using the adonis function. The four sites with significant differences in community structure all contained a site average pH ≤ 5.7 or pH > 6.5 (Supplementary Table 4). Additionally, differential abundance testing of the 74 experimental units from the four sites identified a similar number of enriched ASVs. However, the 282 ASVs, comprised of 77 unique families, enriched under moderate disturbance compared to intense disturbance accounted for similar relative abundances in the two treatments. Like minimum-intense and minimum-moderate comparisons, alpha diversity and richness did not significantly differ ($p > 0.10$) between moderate and intense treatments (Supplementary Fig. 4).

3.3. Modeling respiration with bacterial and archaeal community members

Three hundred and twenty-eight ASVs remained in the respiration model training dataset, post pruning, and on average, accounted for 44% of sequences in rarified experimental units. The respiration model had substantial predictive power ($R^2 = 0.58$) between predicted and actual respiration values. Amplicon sequence variants belonging to *Proteobacteria* contributed the most to variable importance, followed by *Acidobacteria*, and *Verrococcinobacteria*, (Supplementary Fig. 5, Supplementary Table 5). Of the 328 ASVs included in the respiration model, 90 were significantly enriched in minimum disturbance systems compared to intensive disturbance systems (Supplementary Table 6). Additionally, 44% of the ASVs with model importance in the top decile were enriched in minimum disturbance systems compared to intensive disturbance systems (Fig. 5). Furthermore, all but one ASV contained in the top decile of important sequences contained weak, but significant relationships with Cmin ($p < 0.01$), which were identified through individual

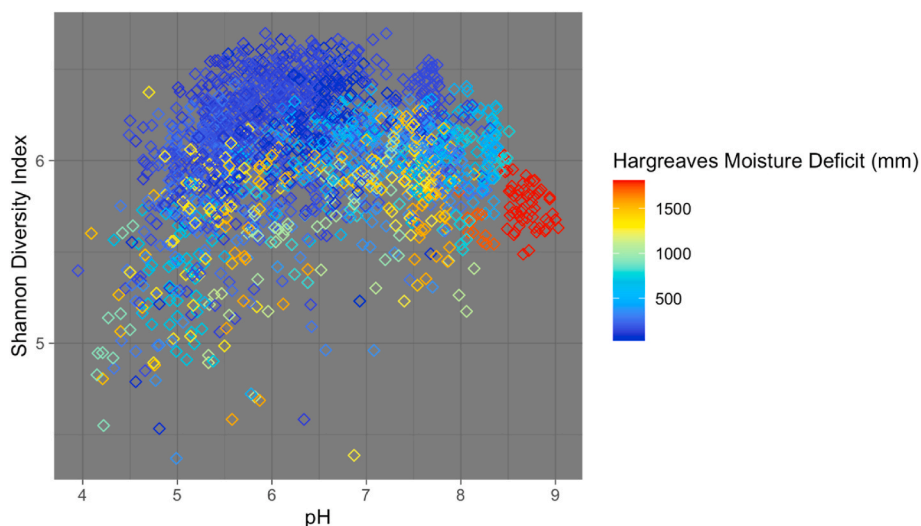


Fig. 2. 16S rRNA Shannon Diversity Index values for North American Project to Evaluate Soil Health Measurements experimental units plotted against pH. Colors represent Hargreaves Moisture Deficit values for each experimental unit. (For interpretation of the references to color in this figure legend, the reader is referred to the Web version of this article.)

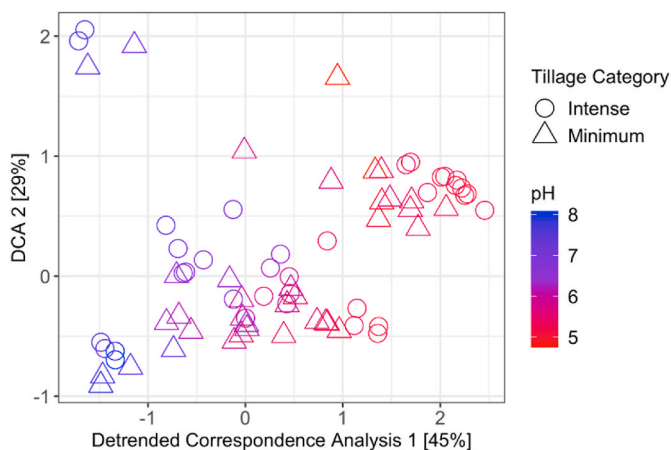


Fig. 3. Detrended correspondence analysis (DCA) of treatments from sites with significant differences in bacterial and archaeal community structure between minimum and intensive disturbance comparisons.

linear regressions, with r^2 values ranging from 0.01 to 0.15. The vast majority of ASVs that were enriched in minimum disturbance and contained within the top decile of model importance contained significant ($p < 0.01$), positive relationships with Cmin (Fig. 5).

4. Discussion

The majority of sites analyzed for shifts in community structure in relation to minimal and intensive disturbance comparisons exhibited significantly different shifts in community structures. The three locations with non-significant differences in community structure had predominantly wheat-based rotations. Previous site-specific studies consistently reported significant differences in community structure because of physical disturbance in corn-based rotations (De Quadros et al., 2012; Sengupta and Dick, 2015; Smith et al., 2016; Srouf et al., 2020). However, prior results from wheat-based rotations are less definitive, with differences in community structure varying by location (Essel et al., 2019; Ng et al., 2012; Schlatter et al., 2019; Yin et al., 2010). Insignificant differences in microbial community structure due to disturbance in wheat-based rotations may stem from greater root density and/or composition associated with wheat plant roots compared to

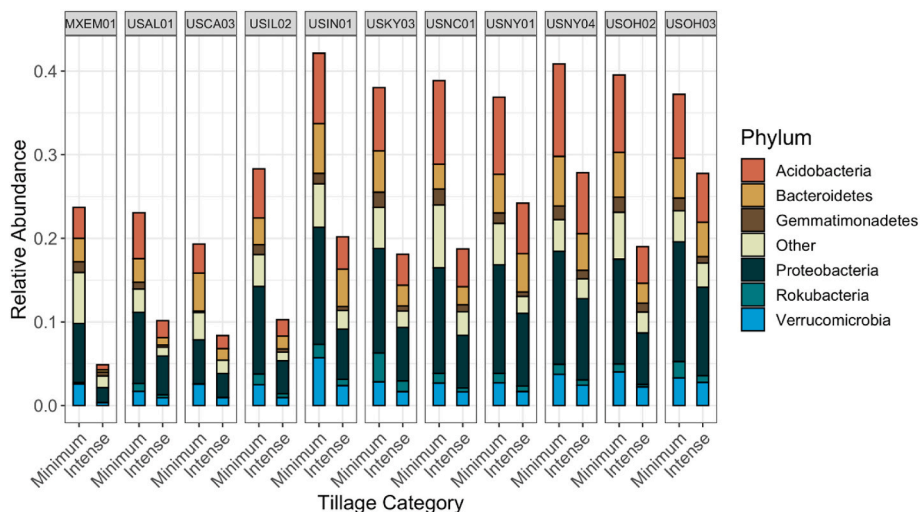


Fig. 4. Average relative abundances of 717 amplicon sequence variants enriched under minimum tillage when compared to intensive tillage, grouped by site.

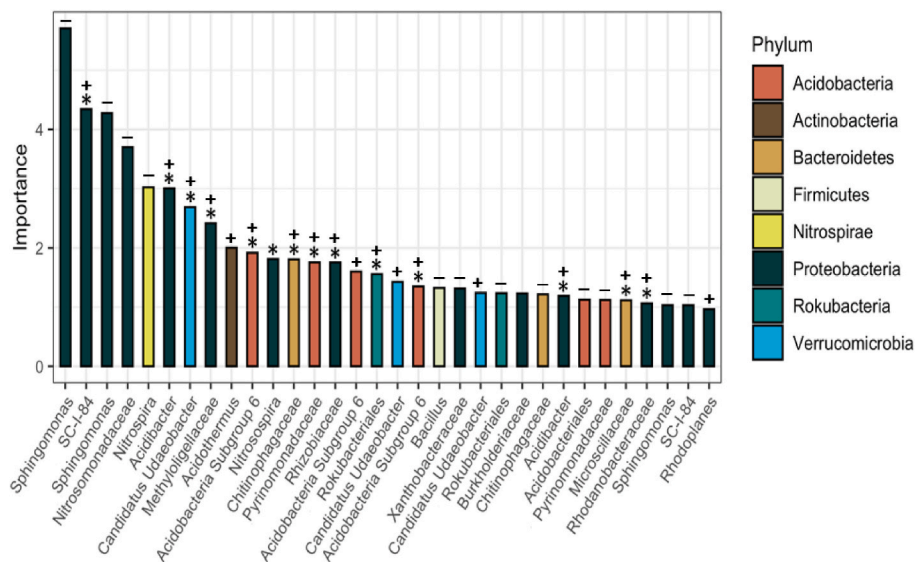


Fig. 5. Top 10% of most important amplicon sequence variants included in the random forest regression potential carbon mineralization model. Sequences are labeled by finest level of available taxonomic classification. Importance was calculated as the average reduction in mean square error across thirty model simulations. Asterisks above a given sequence importance indicates the sequence was enriched under minimum disturbance when compared to intensively disturbed systems. Plus symbols above a given sequence indicate the sequence had a significant ($p < 0.01$), positive relationship with potential carbon mineralization. Negative symbols above a given sequence indicate the sequence had a significant ($p < 0.01$), negative relationship with potential carbon mineralization. Relationships were identified using linear regression.

other crop rooting systems included in the study (Yamaguchi and Tanaka, 1990). Increasing below ground biomass decay may enhance microbial access to nutrient rich organics in a similar fashion to residue incorporation following tillage operations.

Significant differences in minimum-moderate and moderate-intense disturbance comparisons were less consistent in this study, with just over half containing significant differences in community structure due to differences in disturbance treatments. Differences in physical soil disturbance between minimum-moderate disturbance and moderate-intense disturbance treatment comparisons were not as extreme as minimum-intense comparisons. However, trends in significance between these treatments were identified in relation to pH. Minimum-moderate and moderate-intense site level disturbance comparisons did not contain significantly different ($p > 0.01$) microbial community structures at locations where site average pH ranged from 5.7 to 6.5, while locations with site average pH ≤ 5.7 or pH > 6.5 contained significantly different community compositions ($p < 0.01$). Average alpha diversity, measured as Shannon's Diversity Index, and richness were slightly greater at tillage sites with pH ranges from 5.7 to 6.5. Increasing diversity has shown to enhance resilience in some microbial systems, but not in others (Shade et al., 2012). Resiliency in microbial communities is defined as "the rate at which a microbial community returns to its original composition after being disturbed" (Allison and Martiny, 2008). The insignificant differences between less disruptive comparisons are indicative of microbial resilience to physical disruption in slightly acidic soils. This differential response to moderate changes in disturbance when grouped by pH, may be indicative of a link between archaeal and bacterial diversity and resilience driven by inherent features and part of the reason why some soils are less susceptible to changes in function as a result of changes in management (i.e., soils below pH 5.7 or above 6.5). However, this resilience to changes in management may be less evident in soil fungal communities, which are less dependent on soil pH when compared to bacterial communities (Laubert et al., 2008).

The sampling strategy employed allowed us to identify management conditions capable of creating divergent soil microbial community structures. Significant differences in bacterial and archaeal community structure among non-wheat based, minimum-intense tillage comparisons demonstrate community divergence as a result of repeated tillage over multiple years. Differential abundance testing of minimum-intense tillage treatments with significant differences allowed us to identify ASVs responsible for the change in community structure across locations. ASVs identified as *Pedospaerales*, contained within the phylum

Verrucomicrobia, were highly abundant and enriched under minimum tillage conditions when compared to intensive tillage. While *Verrucomicrobia* is a dominant phylum in soil (Bergmann et al., 2011), accounting for over 50% of bacterial 16S rRNA gene sequences in native tall grass prairie soils in the USA (Fierer et al., 2013), their functioning in soil remains poorly understood. Although much remains to be uncovered regarding *Pedospaerales* functional potential, the order has been identified as an indicator of large macroaggregates (Bach et al., 2018), as well as enriched in permanent raised beds when compared to tilled raised beds (Jiménez-Bueno et al., 2016). Coupling indicators of soil health and relative abundance of microbial community members can shed light on the mechanisms behind why practices enhance soils' ability to function. Work has shown that minimum tillage systems compared to intensive tillage generally: 1) increase macroaggregate stability (Al-Kaisi et al., 2014; Zhang et al., 2018), 2) increase soil organic carbon (Nunes et al., 2020), and 3) enhance relative abundance of *Pedospaerales*. Furthermore, tall grass prairies and other minimally managed perennials contain greater measures of aggregate stability and soil organic carbon than row cropping systems that utilize soil health promoting practices (i.e., reduced tillage, cover crops, organic amendments) (Grandy and Robertson, 2007). Together this indicates that in the top 15 cm of soil the repeated pulse events of tillage shift the microbial community and soil structure away from organisms known to exist in unmanaged, natural systems. Conversely a major reduction in physical disturbance can redirect the community structure, back toward a naturally functioning soil.

Reducing tillage often leads to increases in stable aggregates (Al-Kaisi et al., 2014; Zhang et al., 2018). Stable aggregates form niche microbial communities, capable of supporting oligotrophic lifestyles (Bach et al., 2018). ASVs matching *Acidobacteria Subgroup 6* were highly abundant, enriched under minimum disturbance, and important sequences in modeling Cmin. *Acidobacteria* are present in a wide range of habitats across the globe and constitute on average 20% of bacteria in soils (Janssen, 2006). Although *Acidobacteria* are present across a variety of environments, relatively little is known about the phylum due to difficulties related to culturing individual isolates. The first *Subgroup 6* isolates were cultivated in 2011 and characterized as slow growing, adaptive to very low nutrient concentrations, and produced uncharacterized extracellular polymeric substances (George et al., 2011). Sequencing efforts of *Acidobacteria Subgroup 6* have revealed the presence of cellulose synthesis genes and a multitude of high molecular weight proteins with excretion pathway motifs, which are postulated to be involved in extracellular polymeric substance production (Kielak

et al., 2016). Extracellular polymeric substances are largely responsible for building biofilm structure and function (Wingender et al., 1999) and are highly correlated with aggregate stability (Redmile-Gordon et al., 2020). Additionally, extracellular polymeric substance production helps regulate osmotic pressure faced by bacteria under changing moisture regimes (Roberson and Firestone, 1992). Bacteria and archaea capable of producing extracellular polymeric substances have demonstrated higher survival rates when subjected to desiccation (Anderson et al., 2012; Tamaru et al., 2005), which may indicate they are better equipped to resume activity following the drying and rewetting in Cmin measurements.

Candidatus Udaeobacter, contained in the phylum *Verrucomicrobia*, was also enriched under minimum disturbance conditions and was an important contributor to the Cmin regression model. *Candidatus Udaeobacter* is widespread in soils, but has not yet been successfully grown in the laboratory (Poehlein and Schöning, 2020). However, a recently published metagenome-assembled genome revealed enriched amino acid transporter and protease gene concentrations in *Candidatus Udaeobacter* compared to other soil bacteria with larger genomes (Brewer et al., 2016). The authors speculated the bacteria is able to prosper in resource limiting conditions through acquisition of amino acids and vitamins contained in the soil environment, rather than internal biosynthesis (Brewer et al., 2016). One proposed mechanism for Cmin bursts measured following drying and rewetting of soil is consumption of cytoplasmic substances exuded by bacteria to regulate changing osmotic pressure upon rewetting (Fierer and Schimel, 2003). *Candidatus Udaeobacter* and other bacteria capable of utilizing expelled cytoplasmic substances may contribute to greater standardized Cmin measurements in minimum tillage systems when compared to an intensively tilled soil.

Results from NAPESHM indicated Cmin was significantly greater in systems managed for minimal physical disturbance when compared to systems with greater physical disturbance (Liptzin et al., n.d.). However, these results are not indicative of *in situ* carbon mineralization in similar cropping systems where mineralization is lower in systems managed for minimal disturbance (Abdalla et al., 2016). Potential carbon mineralization measured in the laboratory rewetting incubation is a result of the consumption of cells lysed during drying, fresh metabolic waste, and newly available organic residues resulting from physical disruption (Fierer and Schimel, 2003; Kaiser et al., 2015). Soils managed for minimal physical disturbance generally contain greater amounts of organic carbon in the top 15 cm than highly disturbed counterparts (Nunes et al., 2020). Increases in organic carbon may be partially attributed to increases in aggregation, which can lead to organic residues becoming physically unavailable for microbial consumption (Paustian et al., 2019). Disruption of aggregates through sieving prior to the incubation releases an unknown fraction of organic residues previously unavailable for microbial consumption. The diverse set of bacterial and archaeal taxa whose abundances were important in predicting Cmin are indicative of the broad range of newly available organic compounds available for microbial consumption. Furthermore, within the top 15 cm of soil, many bacterial and archaeal community members reside in aggregates containing unique sets of organisms, which adapt to organic matter resources, pore-space networks, and water and oxygen availability (Bach et al., 2018). Disruption of these habitats during the sieving and drying process may alter access to water, oxygen, and organic matter resources, depending on the soil in question. Understanding which bacteria and archaea are capable of thriving post drying and sieving and the mechanisms they employ (e.g., biofilm formation, alternative nutrient consumption) will provide further context as to why soils managed for minimum disturbance report greater Cmin than their disturbed counterparts. Sequences deemed important when modeling Cmin and enriched under minimum disturbance provide a starting point for understanding the microbial community members which influence Cmin.

4.1. Conclusions

The unique sampling design incorporated in this study elucidated a set of ASVs that were enriched in soils under minimum tillage management across soil types and climates. Results from this study indicate that type of cropping system, intensity of physical disruption, and soil pH all influence the degree of archaeal and bacterial sensitivity to tillage. The multiple factors capable of affecting bacterial and archaeal community structure may explain divergent results among site specific studies and underscore the importance of limiting interpolation of individual studies to a broad scale. These results indicate a benefit to performing analyses across a range of agricultural soils to capture how core microbial members respond to changes in agricultural management.

The subset of ASVs enriched under minimum disturbance management and identified as important variables in random forest regression modeling, provide insight into the bacterial and archaeal community members who may be responsible for increases in Cmin measurements in systems with reduced tillage. While many biologically based soil health measurements are sensitive to management, few are tied to changes in soil function. Better understanding the microbial drivers of widely used soil health indicators, such as Cmin, rather than interpreting increases in measurements as an indication of increasing soil health, provides stakeholders context as to why the measures increase in soils managed for minimal disturbance. Soils with reduced disturbance foster bacterial and archaeal organisms with diverse lifestyles as a function of niche diversity developed within stable aggregates. Stable microbial communities formed in aggregates in soils managed for minimal physical disturbance represent communities capable of functioning in the absence of nutrient rich amendments. Understanding how soil microorganisms adapt and function in agricultural systems managed for minimal physical disturbance may provide the appropriate context for stakeholders to interpret Cmin results and help guide adoption of the practice.

Funding information

Foundation for Food and Agricultural Research (grant ID 523926), General Mills, and The Samuel Roberts Noble Foundation.

Declaration of competing interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

Acknowledgements

The NAPESHM project is part of a broader effort titled, "Assessing and Expanding Soil Health for Production, Economic, and Environmental Benefits". The project is funded by the Foundation for Food and Agricultural Research (grant ID 523926), General Mills, and The Samuel Roberts Noble Foundation. The content of this publication is solely the responsibility of the authors and does not necessarily represent the official views of the Foundation for Food and Agriculture Research. The authors acknowledge the following individuals for their contribution to the long-term research sites Rob Dungan, Paul DeMaere, Merf Solorio, Jaime Solorio, Tracy Waltrip, Nelson Vallejo, Mark Strole, Bob Blevens, George Kapusta, Ronald Krausz, Karla Gage, Rachel Cook, Amanda Weidhuner, Con Campbell, Brian McConkey, Harold van Es, Hong Wang, Reynald Lemke, Kelsey Brandt, D. Lawrence, M.R. Reebe, B. Horner, M. Sultani, Sean Vink, Mike Zink, Melissa Bell, Nancy Creamer, Alan Franzluebbers, Tomas Moreno, Paul Mueller, Chris Reberg-Horton, Joshua Heitman, April Leytem, Mark Liebig, Deanna Osmond, Michael Thompson, Martin Entz, and K. Rinas. Thank you.

Appendix A. Supplementary data

Supplementary data to this article can be found online at <https://doi.org/10.1016/j.soilbio.2022.108618>.

References

- Abdalla, K., Chivenge, P., Ciaes, P., Chaplot, V., 2016. No-tillage lessens soil CO₂ emissions the most under arid and sandy soil conditions: results from a meta-analysis. *Biogeosciences* 13, 3619–3633. <https://doi.org/10.5194/BG-13-3619-2016>.
- Acosta-Martinez, V., Cano, A., Johnson, J., 2018. Simultaneous determination of multiple soil enzyme activities for soil health-biogeochemical indices. *Applied Soil Ecology* 126, 121–128. <https://doi.org/10.1016/j.apsoil.2017.11.024>.
- Al-Kaisi, M.M., Douelle, A., Kwaw-Mensah, D., 2014. Soil microaggregate and macroaggregate decay over time and soil carbon change as influenced by different tillage systems. *Journal of Soil and Water Conservation* 69, 574–580. <https://doi.org/10.2489/JSWC.69.6.574>.
- Allison, S.D., Martiny, J.B.H., 2008. Resistance, resilience, and redundancy in microbial communities. *Proceedings of the National Academy of Sciences of the United States of America* 105, 11512–11519. <https://doi.org/10.1073/pnas.0801925105>.
- Anderson, K.L., Apolinario, E.E., Sowers, K.R., 2012. Desiccation as a long-term survival mechanism for the archaeon *Methanosarcina barkeri*. *Applied and Environmental Microbiology* 78, 1473–1479. <https://doi.org/10.1128/AEM.06964-11>.
- Andrews, S.S., Karlen, D.L., Cambardella, C.A., 2004. The soil management assessment framework: a quantitative soil quality evaluation method. *Soil Science Society of America Journal* 68, 1945–1962.
- Bach, E.M., Williams, R.J., Hargreaves, S.K., Yang, F., Hofmockel, K.S., 2018. Greatest soil microbial diversity found in micro-habitats. *Soil Biology and Biochemistry* 118, 217–226. <https://doi.org/10.1016/j.soilbio.2017.12.018>.
- Bergmann, G.T., Bates, S.T., Eilers, K.G., Lauber, C.L., Caporaso, J.G., Walters, W.A., Knight, R., Fierer, N., 2011. The under-recognized dominance of Verrucomicrobia in soil bacterial communities. *Soil Biology and Biochemistry* 43, 1450–1455. <https://doi.org/10.1016/j.soilbio.2011.03.012>.
- Brewer, T.E., Handley, K.M., Carini, P., Gilbert, J.A., Fierer, N., 2016. Genome reduction in an abundant and ubiquitous soil bacterium “*Candidatus Udaobacter copiosus*”. *Nature Microbiology* 2, 1–7. <https://doi.org/10.1038/nmicrobiol.2016.198>.
- Campbell, G.S., Jackson, R.D., Mortland, M.M., Nielsen, D.R., Klute, A., 2018. Methods of soil analysis. *Methods of Soil Analysis, Part 1: Physical and Mineralogical Methods* 1–1188. <https://doi.org/10.2136/SSSABOOKSER5.1.2ED>.
- Caporaso, J.G., Ackermann, G., Apprill, A., Bauer, M., Berg-Lyons, D., Betley, J., Fierer, N., Fraser, L., Fuhrman, J.A., Gilbert, J.A., Gormley, N., Humphrey, G., Huntley, J., Jansson, J.K., Knight, R., Lauber, C.L., Lozupone, C.A., Mcnally, S., Needham, D.M., Owens, S.M., Parada, A.E., Parsons, R., Smith, G., Thompson, L.R., Thompson, L., Turnbaugh, P.J., Walters, W.A., Weber, L., 2018. EMP 16S Illumina amplicon protocol. *PLoS One*. <https://doi.org/10.17504/protocols.io.nuudeww>.
- Ceja-Navarro, J.A., Rivera-Orduña, F.N., Patiño-Zúñiga, L., Vila-Sanjurjo, A., Crossa, J., Govaerts, B., Dendooven, L., 2010. Phylogenetic and multivariate analyses to determine the effects of different tillage and residue management practices on soil bacterial communities. *Applied and Environmental Microbiology* 76, 3685–3691. <https://doi.org/10.1128/AEM.02726-09>.
- Culman, S.W., Snapp, S.S., Freeman, A.J., Schipanski, M.E., Beniston, J., Lal, R., Drinkwater, L.E., Franzluebbers, A.J., Glover, J.D., Grandy, A.S., Lee, J., Six, J., Maul, J.E., Mirsky, S.B., Spargo, J.T., Wander, M.M., 2012. Permanganate oxidizable carbon reflects a processed soil fraction that is sensitive to management. *Soil Science Society of America Journal* 76, 494–504. <https://doi.org/10.2136/sssaj2011.0286>.
- De Quadros, P.D., Zhalnina, K., Davis-Richardson, A., Fagen, J.R., Drew, J., Bayer, C., Camargo, F.A.O., Triplett, E.W., 2012. The effect of tillage system and crop rotation on soil microbial diversity and composition in a subtropical acrisol. *Diversity* 4, 375–395. <https://doi.org/10.3390/d4040375>.
- Dixon, P., 2003. VEGAN, a package of R functions for community ecology. *Journal of Vegetation Science* 14, 927–930. <https://doi.org/10.1111/j.1654-1103.2003.tb02228.x>.
- Dreimanis, A., 1962. Quantitative gasometric determination of calcite and dolomite by using chittick apparatus. *SEPM Journal of Sedimentary Research* 32, 520–529. <https://doi.org/10.1306/74d70d08-2b21-11d7-8648000102c1865d>.
- Essel, E., Xie, Junhong, Deng, C., Peng, Z., Wang, J., Shen, J., Xie, Jianhui, Coulter, J.A., Li, L., 2019. Bacterial and fungal diversity in rhizosphere and bulk soil under different long-term tillage and cereal/legume rotation. *Soil and Tillage Research* 194, 104302. <https://doi.org/10.1016/j.still.2019.104302>.
- Fierer, N., Ladau, J., Clemente, J.C., Leff, J.W., Owens, S.M., Pollard, K.S., Knight, R., Gilbert, J.A., McCulley, R.L., 2013. Reconstructing the microbial diversity and function of pre-agricultural tallgrass prairie soils in the United States. *Science* 342, 621–624. <https://doi.org/10.1126/science.1243768>.
- Fierer, N., Schimel, J.P., 2003. A proposed mechanism for the pulse in carbon dioxide production commonly observed following the rapid rewetting of a dry soil. *Soil Science Society of America Journal* 67, 798–805. <https://doi.org/10.2136/sssaj2003.7980>.
- Fierer, N., Wood, S.A., Bueno de Mesquita, C.P., 2021. How microbes can, and cannot, be used to assess soil health. *Soil Biology and Biochemistry* 153, 108111. <https://doi.org/10.1016/j.soilbio.2020.108111>.
- Gee, G.W., Or, D., 2018. 2.4 Particle-Size Analysis. *John Wiley & Sons, Ltd*, pp. 255–293. <https://doi.org/10.2136/sssabookser5.4.c12>.
- George, I.F., Hartmann, M., Liles, M.R., Agathos, S.N., 2011. Recovery of as-yet-uncultured soil Acidobacteria on dilute solid media. *Applied and Environmental Microbiology* 77, 8184–8188. <https://doi.org/10.1128/AEM.05956-11>.
- Gonzalez-Quinones, V., Stockdale, E.A., Banning, N.C., Hoyle, F.C., Sawada, Y., Wherrett, A.D., Jones, D.L., Murphy, D.V., 2011. Soil microbial biomass-Interpretation and consideration for soil monitoring. *Soil Research* 49, 287–304. <https://doi.org/10.1071/SR10203>.
- Grandy, A.S., Robertson, G.P., 2007. Land-use intensity effects on soil organic carbon accumulation rates and mechanisms. *Ecosystems* 10 (1 10), 59–74. <https://doi.org/10.1007/S10021-006-9010-Y>, 2007.
- Guo, Z., Han, J., Li, J., Xu, Y., Wang, X., 2019. Effects of long-term fertilization on soil organic carbon mineralization and microbial community structure. *PLoS One* 14, e0211163. <https://doi.org/10.1371/JOURNAL.PONE.0211163>.
- Haney, R.L., Haney, E.B., Hossner, L.R., Arnold, J.G., 2010. Modifications to the new soil extractant H3A-1: a multnutrient extractant. *Communications in Soil Science and Plant Analysis* 41, 1513–1523. <https://doi.org/10.1080/00103624.2010.482173>.
- Haney, R.L., Haney, E.B., Smith, D.R., Harmel, R.D., White, M.J., 2018. The soil health tool—theory and initial broad-scale application. *Applied Soil Ecology* 125, 162–168. <https://doi.org/10.1016/j.apsoil.2017.07.035>.
- Hargreaves, G.H., Allen, R.G., 2003. History and evaluation of Hargreaves evapotranspiration equation. *Journal of Irrigation and Drainage Engineering* 129, 53–63. [https://doi.org/10.1061/\(asce\)0733-9437\(2003\)129:1\(53\)](https://doi.org/10.1061/(asce)0733-9437(2003)129:1(53)).
- Janssen, P.H., 2006. Identifying the dominant soil bacterial taxa in libraries of 16S rRNA and 16S rRNA genes. *Applied and Environmental Microbiology* 72, 1719–1728. <https://doi.org/10.1128/AEM.72.3.1719-1728.2006>.
- Jiménez-Bueno, N.G., Valenzuela-Encinas, C., Marsch, R., Ortiz-Gutiérrez, D., Verhulst, N., Govaerts, B., Dendooven, L., Navarro-Noya, Y.E., 2016. Bacterial indicator taxa in soils under different long-term agricultural management. *Journal of Applied Microbiology* 120, 921–933. <https://doi.org/10.1111/jam.13072>.
- Kaiser, M., Kleber, M., Berhe, A.A., 2015. How air-drying and rewetting modify soil organic matter characteristics: an assessment to improve data interpretation and inference. *Soil Biology and Biochemistry*. <https://doi.org/10.1016/j.soilbio.2014.10.018>.
- Khanghahi, M.Y., Murgese, P., Strafella, S., Crecchio, C., 2019. Soil biological fertility and bacterial community response to land use intensity: a case study in the mediterranean area. *Diversity* 11, 211. <https://doi.org/10.3390/D11110211>.
- Kielak, A.M., Barreto, C.C., Kowalchuk, G.A., van Veen, J.A., Kuramae, E.E., 2016. The ecology of Acidobacteria: moving beyond genes and genomes. *Frontiers in Microbiology*. <https://doi.org/10.3389/fmicb.2016.00744>.
- Lauber, C.L., Strickland, M.S., Bradford, M.A., Fierer, N., 2008. The influence of soil properties on the structure of bacterial and fungal communities across land-use types. *Soil Biology and Biochemistry* 40, 2407–2415. <https://doi.org/10.1016/J.SOILBIO.2008.05.021>.
- Li, C., Cano, A., Acosta-Martinez, V., Veum, K.S., Moore-Kucera, J., 2020. A comparison between fatty acid methyl ester profiling methods (PLFA and EL-FAME) as soil health indicators. *Soil Science Society of America Journal* 84, 1153–1169. <https://doi.org/10.1002/saj2.20118>.
- Liptzin, D., Norris, C.E., Cappellazzi, S.B., Bean, G. Mac, Cope, M., Greub, K.L.H., Rieke, E.L., Tracy, P.W., Morgan, C.L.S., Honeycutt, C.W., n.d. An evaluation of carbon indicators of soil health in long-term agricultural experiments. *Soil Biology and Biochemistry*.
- Liu, Y.R., Delgado-Baquerizo, M., Wang, J.T., Hu, H.W., Yang, Z., He, J.Z., 2018. New insights into the role of microbial community composition in driving soil respiration rates. *Soil Biology and Biochemistry* 118, 35–41. <https://doi.org/10.1016/J.SOILBIO.2017.12.003>.
- Love, M., Anders, S., Huber, W., Love, M.M., 2018. Package “DESeq2”. <https://doi.org/10.1186/s13059-014-0550-8>.
- Malik, A.A., Puissant, J., Buckeridge, K.M., Goodall, T., Jehmlich, N., Chowdhury, S., Gweon, H.S., Peyton, J.M., Mason, K.E., van Agtmaal, M., Blaud, A., Clark, I.M., Whitaker, J., Pywell, R.F., Ostle, N., Gleixner, G., Griffiths, R.I., 2018. Land use driven change in soil pH affects microbial carbon cycling processes. *Nature Communications* 9 (1 9), 1–10. <https://doi.org/10.1038/s41467-018-05980-1>, 2018.
- Mbuthia, L.W., Acosta-Martínez, V., DeBryun, J., Schaeffer, S., Tyler, D., Odoi, E., Mpheshea, M., Walker, F., Eash, N., 2015. Long term tillage, cover crop, and fertilization effects on microbial community structure, activity: implications for soil quality. *Soil Biology and Biochemistry* 89, 24–34. <https://doi.org/10.1016/J.SOILBIO.2015.06.016>.
- McMurdie, P.J., Holmes, S., 2013. phyloseq: an R package for reproducible interactive analysis and graphics of microbiome census data. *PLoS One* 8, e61217. <https://doi.org/10.1371/journal.pone.0061217>.
- Moebius-Clune, B., Moebius-Clune, D.J., Gugino, B., Idowu, O., Schindelbeck, R., Ristow, A., van Es, H., Thies, J., Shayler, H.A., McBride, M.B., Wolfe, D., Abawi, G., 2016. *Comprehensive Assessment of Soil Health—the Cornell Framework Manual*, 3.1. Cornell University, Geneva, NY.
- Navarro-Noya, Y.E., Gómez-Acata, S., Montoya-Ciriaco, N., Rojas-Valdez, A., Suárez-Arriaga, M.C., Valenzuela-Encinas, C., Jiménez-Bueno, N., Verhulst, N., Govaerts, B., Dendooven, L., 2013. Relative impacts of tillage, residue management and crop-rotation on soil bacterial communities in a semi-arid agroecosystem. *Soil Biology and Biochemistry* 65, 86–95. <https://doi.org/10.1016/j.soilbio.2013.05.009>.
- Nelson, D.W., Sommers, L.E., 2015. *Total Carbon, Organic Carbon, and Organic Matter*. John Wiley & Sons, Ltd, pp. 539–579. <https://doi.org/10.2134/agronmonogr9.2.2ed.c29>.
- 2012, U Ng J., Hollister, E., Ecology, M.G.-C.-I., 2012. Impacts of cropping systems and long-term tillage on soil microbial population levels and community composition in

- dryland agricultural setting. *ISRN Ecology* 1–11. <https://doi.org/10.5402/2012/487370>, 2012.
- Norris, C.E., Bean, G. Mac, Cappellazzi, S.B., Cope, M., Greub, K.L.H., Liptzin, D., Rieke, E.L., Tracy, P.W., Morgan, C.L.S., Honeycutt, C.W., 2020. Introducing the North American project to evaluate soil health measurements. *Agronomy Journal* 112, 3195–3215. <https://doi.org/10.1002/agj2.20234>.
- Nunes, M.R., Karlen, D.L., Veum, K.S., Moorman, T.B., Cambardella, C.A., 2020. Biological soil health indicators respond to tillage intensity: a US meta-analysis. *Geoderma* 369, 114335. <https://doi.org/10.1016/j.geoderma.2020.114335>.
- Paustian, K., Collins, H.P., Paul, E.A., 2019. Management controls on soil carbon. *Soil Organic Matter in Temperate Agroecosystems* 15–49. <https://doi.org/10.1201/9780367811693-2>.
- Pedregosa, F., Varoquaux, G., Gramfort, A., Michel, V., Thirion, B., Grisel, O., Blondel, M., Prettenhofer, P., Weiss, R., Dubourg, V., Vanderplas, J., Passos, A., Cournapeau, D., 2011. Scikit-learn: machine learning in Python. *Journal of Machine Learning Research* 12, 2825–2830.
- Poehlein, A., Schöning, I., 2020. Globally Abundant “Candidatus Udaebacter” Benefits from Release of Antibiotics in Soil and Potentially Performs Trace Gas Scavenging. <https://doi.org/10.1128/mSphere.00186-20>.
- Redmile-Gordon, M., Gregory, A.S., White, R.P., Watts, C.W., 2020. Soil organic carbon, extracellular polymeric substances (EPS), and soil structural stability as affected by previous and current land-use. *Geoderma* 363, 114143. <https://doi.org/10.1016/j.geoderma.2019.114143>.
- Roberson, E.B., Firestone, M.K., 1992. Relationship between desiccation and exopolysaccharide production in a soil *Pseudomonas* sp. *Applied and Environmental Microbiology* 58, 1284–1291. <https://doi.org/10.1128/aem.58.4.1284-1291.1992>.
- Schlatter, D.C., Paul, N.C., Shah, D.H., Schillinger, W.F., Bary, A.I., Sharratt, B., Paulitz, T.C., 2019. Biosolids and tillage practices influence soil bacterial communities in dryland wheat. *Microbial Ecology* 78, 737–752. <https://doi.org/10.1007/s00248-019-01339-1>.
- Schmidt, R., Gravuer, K., Bossange, A.V., Mitchell, J., Scow, K., 2018. Long-term use of cover crops and no-till shift soil microbial community life strategies in agricultural soil. *PLoS One* 13, e0192953. <https://doi.org/10.1371/JOURNAL.PONE.0192953>.
- Schmidt, R., Mitchell, J., Scow, K., 2019. Cover cropping and no-till increase diversity and symbiotroph:saprotroph ratios of soil fungal communities. *Soil Biology and Biochemistry* 129, 99–109. <https://doi.org/10.1016/J.SOILBIO.2018.11.010>.
- Sengupta, A., Dick, W.A., 2015. Bacterial community diversity in soil under two tillage practices as determined by pyrosequencing. *Microbial Ecology* 70, 853–859. <https://doi.org/10.1007/s00248-015-0609-4>.
- Shade, A., Peter, H., Allison, S.D., Baho, D.L., Berga, M., Bürgmann, H., Huber, D.H., Langenheder, S., Lennon, J.T., Martiny, J.B.H., Matulich, K.L., Schmidt, T.M., Handelsman, J., 2012. Fundamentals of microbial community resistance and resilience. *Frontiers in Microbiology*. <https://doi.org/10.3389/fmicb.2012.00417>.
- Smith, C.R., Blair, P.L., Boyd, C., Cody, B., Hazel, A., Hedrick, A., Kathuria, H., Khurana, P., Kramer, B., Muterspaw, K., Peck, C., Sells, E., Skinner, J., Tegeler, C., Wolfe, Z., 2016. Microbial community responses to soil tillage and crop rotation in a corn/soybean agroecosystem. *Ecology and Evolution* 6, 8075–8084. <https://doi.org/10.1002/ece3.2553>.
- Srour, A.Y., Ammar, H.A., Subedi, A., Pimentel, M., Cook, R.L., Bond, J., Fakhoury, A.M., 2020. Microbial communities associated with long-term tillage and fertility treatments in a corn-soybean cropping system. *Frontiers in Microbiology* 11, 1363. <https://doi.org/10.3389/fmicb.2020.01363>.
- Tamaru, Y., Takani, Y., Yoshida, T., Sakamoto, T., 2005. Crucial role of extracellular polysaccharides in desiccation and freezing tolerance in the terrestrial cyanobacterium *Nostoc commune*. *Applied and Environmental Microbiology* 71, 7327–7333. <https://doi.org/10.1128/AEM.71.11.7327-7333.2005>.
- Wang, T., Hamann, A., Spittlehouse, D., Carroll, C., 2016. Locally downscaled and spatially customizable climate data for historical and future periods for North America. *PLoS One* 11, e0156720. <https://doi.org/10.1371/journal.pone.0156720>.
- Wang, Z., Liu, L., Chen, Q., Wen, X., Liao, Y., 2016. Conservation tillage increases soil bacterial diversity in the dryland of northern China. *Agronomy for Sustainable Development* 36, 1–9. <https://doi.org/10.1007/s13593-016-0366-x>.
- Weiss, S., Xu, Z.Z., Peddada, S., Amir, A., Bittinger, K., Gonzalez, A., Lozupone, C., Zaneveld, J.R., Vázquez-Baeza, Y., Birmingham, A., Hyde, E.R., Knight, R., 2017. Normalization and microbial differential abundance strategies depend upon data characteristics. *Microbiome* 1–18. <https://doi.org/10.1186/S40168-017-0237-Y>, 2017 5:1 5.
- Wingender, J., Neu, T.R., Flemming, H.-C., 1999. What are bacterial extracellular polymeric substances? In: *Microbial Extracellular Polymeric Substances*. Springer Berlin Heidelberg, pp. 1–19. https://doi.org/10.1007/978-3-642-60147-7_1.
- Yamaguchi, J., Tanaka, A., 1990. Quantitative observation on the root system of various crops growing in the field. *Soil Science & Plant Nutrition* 36, 483–493. <https://doi.org/10.1080/00380768.1990.10416917>.
- Yin, C., Jones, K.L., Peterson, D.E., Garrett, K.A., Hulbert, S.H., Paulitz, T.C., 2010. Members of soil bacterial communities sensitive to tillage and crop rotation. *Soil Biology and Biochemistry* 42, 2111–2118. <https://doi.org/10.1016/j.soilbio.2010.08.006>.
- Zhang, X., Xin, X., Zhu, A., Yang, W., Zhang, J., Ding, S., Mu, L., Shao, L., 2018. Linking macroaggregation to soil microbial community and organic carbon accumulation under different tillage and residue managements. *Soil and Tillage Research* 178, 99–107. <https://doi.org/10.1016/J.STILL.2017.12.020>.
- Zibilske, L.M., 2018. Carbon mineralization. *Environmental Science and Technology* 30, 835–863. <https://doi.org/10.2136/sssabookser5.2.c38>.