

RESEARCH LETTER

High rhizospheric ammonium levels in *Sorghum halepense* (johnsongrass) suggests nitrification inhibition potential

Eeshita Ghosh | Nithya Rajan  | Dinesh Phuyal | Nithya Subramanian | Muthukumar Bagavathiannan 

Department of Soil and Crop Sciences,
Texas A&M University, College Station,
Texas, USA

Correspondence

Muthukumar Bagavathiannan, Department
of Soil and Crop Sciences, Texas A&M
University, College Station, TX, USA.
Email: Muthu.bagavathiannan@tamu.edu

Assigned to Associate Editor Amitava
Chatterjee.

Funding information

USDA-NIFA Foundational Project

Abstract

Plants, such as sorghum (*Sorghum bicolor*), have been shown to secrete root exudates involved in biological nitrification inhibition (BNI), an ability to suppress the conversion of ammonium to nitrate and thereby minimize its loss. Johnsongrass (*Sorghum halepense*), a weedy relative of cultivated sorghum, may also possess BNI potential, but little is known in this regard. Here, we conducted a field survey at seven different sites in Southeast Texas to determine this evolutionary trait of johnsongrass in different soil environments. It was found that johnsongrass rhizosphere retains high levels (>60%) of ammonium within the total available N (ammonium + nitrate). Furthermore, the degree of ammonium retention by johnsongrass rhizosphere was significantly greater (up to 40%) in the roadside habitat compared to cultivated fields. The high ammonium retention potential by johnsongrass may explain, in part, their persistence and dominance, especially in marginal environments.

1 | INTRODUCTION

Nitrogen (N) is one of the most limiting plant nutrients, which is taken up by plants in the form of ammonium (NH_4^+) or nitrate (NO_3^-) (Vitousek & Howarth, 1991). Ammonium, being positively charged, can adhere to the negatively charged soil particles, whereas negatively charged nitrate is mobile and rapidly lost through leaching or nitrous oxide (Coskun et al., 2017). In the soil, ammonium is actively converted to nitrate by various microorganisms, mainly by ammonia-oxidizing archaea and bacteria (conversion of ammonium to nitrite) and nitrite-oxidizing bacteria (conversion of nitrite to nitrate) (Coskun et al., 2017; Hayatsu et al., 2008; Hu et al., 2014; Kowalchuk & Stephen, 2001). These loss processes greatly reduce N availability for plant growth (Subbarao et al.,

2015), and accumulation of excess N in the soil, air, and water is a global problem, leading to the deposition of reactive N compounds in the environment (Singh & Verma, 2007; Stark & Richards, 2008; Vetsch et al., 2019). This process, in turn, reduces agricultural production efficiency, while contributing to environmental pollution and climate change (Anderson et al., 2020; EPA, 2024; Mueller et al., 2019; Stark & Richards, 2008).

In natural ecosystems, plants may have evolved unique associations/interactions with microbial organisms, compared to the cultivated systems where agricultural inputs and management might differentially influence such interactions (Soldan et al., 2021). The rhizosphere contains a wide range of microbial groups that participate in activities such as nutrient cycling, protection against plant diseases, as well as responding to both biotic and abiotic stress factors. This relationship indicates that the rhizosphere can be utilized and

Abbreviation: BNI, biological nitrification inhibition.

This is an open access article under the terms of the [Creative Commons Attribution](https://creativecommons.org/licenses/by/4.0/) License, which permits use, distribution and reproduction in any medium, provided the original work is properly cited.

© 2024 The Author(s). *Agricultural & Environmental Letters* published by Wiley Periodicals LLC on behalf of American Society of Agronomy, Crop Science Society of America, and Soil Science Society of America.

manipulated to enhance plant growth, nutrient acquisition, tolerance to biotic and abiotic stress factors, and overall productivity (Brisson et al., 2019; Bulgarelli et al., 2015; Hakim et al., 2021).

Some plant species, such as *Brachiaria humidicola* (koronivia grass) (Ishikawa et al., 2003) and *Sorghum bicolor* (sorghum) (Coskun et al., 2017; Subbarao et al., 2013), have evolved mechanisms to inhibit nitrification by secreting root exudates that target the enzymes in different steps of nitrification known as biological nitrification inhibition (BNI) (Subbarao et al., 2006). These root exudates target either ammonia monooxygenase or hydroxylamine oxidoreductase or both, which are enzymes involved in the conversion of ammonia to hydroxylamine (rate-limiting step) and hydroxylamine to nitrite, respectively (Coskun et al., 2017; Subbarao et al., 2013). This allows a plant to retain greater amounts of N in the form of ammonia, which in turn improves its growth. Modern crops have mostly lost these traits and are heavily dependent on human management, requiring substantial fertilizer inputs.

Sorghum halepense (johnsongrass), a weedy relative of cultivated sorghum, is a troublesome and invasive weed species throughout the southern United States (Klein & Smith, 2020; Ohadi et al., 2017). Therefore, the dominance and invasiveness of johnsongrass could be attributed in part to its nitrification inhibition properties by retaining soil N in the form of ammonium for longer periods of time. This ability is particularly important for johnsongrass growing in roadside habitats due to the lack of external fertilization compared to the biotypes in croplands. We hypothesize that the johnsongrass growing in roadside habitats has evolutionary advantages through the retaining of more ammonium in the rhizosphere compared to johnsongrass growing in croplands. However, little has been investigated in this regard. Here, we conducted a field survey in the Southeast Texas region to determine the extent of ammonium retention in naturally occurring johnsongrass biotypes in roadsides versus cropland habitats.

2 | MATERIALS AND METHODS

2.1 | Soil sample collection

A survey was conducted in seven randomly chosen locations (Snook, Mumford, Somerville, Wharton, Cameron, Thrall, and Eagle Lake) in Southeast Texas during late October and November 2021 (Table 1). In each location, a pair of field (cropland) and roadside sites that occurred within a 1 km distance was chosen. These sites were randomly chosen a priori on a Google map, without any specific knowledge/consideration to the site characteristics or the growth habits of johnsongrass. Among these, the field sites of Eagle Lake and Thrall locations were considered a special case since

Core Ideas

- Nitrogen is a limiting nutrient for plant growth, and nitrification causes loss of nitrogen.
- Ammonium retention was higher in roadside johnsongrass biotypes compared to that of cropland biotypes.
- The high rhizospheric ammonium retention by johnsongrass may explain, at least in part, its invasiveness.
- This trait could be further investigated and integrated into modern sorghum cultivars.

they were under very dense johnsongrass infestation. Within each site, 10 johnsongrass plants were randomly selected (at least 2 m apart), and three pooled soil cores (15-cm depth \times 2.5-cm diameter), having three replications each, were collected from the rhizosphere region of each plant. In addition, soil cores were collected in areas without johnsongrass infestation (check treatment). All the rhizosphere soil samples were placed in ice boxes ($\sim 4^{\circ}\text{C}$) immediately after collection and stored in a freezer (-20°C) until further analysis.

2.2 | Soil characterization

To determine the general soil characteristics of the survey site, 16 soil cores (30-cm depth \times 2.5-cm diameter) were collected randomly from each site; soil texture and pH were analyzed at the Texas A&M AgriLife Extension Service Soil, Water, and Forage Testing Laboratory (Table 1). Additionally, plant density/biomass production was visually scored on a scale of 1–10, with 10 indicating the highest possible density and biomass in the site (Table 1). All the johnsongrass plants in the survey sites were taller than 15 cm, except for the field in Wharton, where the plants were in the vegetative growth stage and small. In each location pair (i.e., field vs. roadside), plant density conditions were comparable (except for Snook and Somerville, due to the unavailability of comparable sites within 1 km distance).

2.3 | Soil ammonium and nitrate analysis

To extract ammonium and nitrate from the rhizosphere soil samples, 10 g of the moist soil sample was used. The soil was sieved (2 mm), and the entire soil N content was extracted using 40 mL 0.5 M potassium sulfate (K_2SO_4) as an extractant solution (Mulaney, 1996). Later, the solution was filtered using 0.2- μm filters. A separate subsample of the soil was placed in an oven at 105°C for 24 h to determine soil

TABLE 1 Previous crop, soil type, soil pH, growth stage and plant density of the various sites from where the samples were collected.

Location	Site	Coordinates		Previous crop/setting	pH	Soil type & texture	Growth stage ^a	Plant density/biomass ^b (1–10)
		Latitude (°N)	Longitude (°W)					
Snook	Field	30.553	96.431	Fiber hemp	8.0	Clay loam	Reproductive	3
	Roadside	30.557	96.428	Grass	8.0	Clay	Reproductive	5
Mumford	Field	30.584	96.499	Corn	8.4	Loam	Reproductive	6
	Roadside	30.583	96.495	Grass	7.9	Loam	Vegetative	6
Somerville	Field	30.513	96.430	Corn	7.8	Clay	Reproductive	7
	Roadside	30.514	96.428	Grass	7.6	Clay loam	Flowering	9
Wharton	Field	29.340	96.123	Pasture	7.5	Clay	Vegetative	6
	Roadside	29.334	96.128	Grass	7.6	Clay loam	Reproductive	6
Cameron	Field	30.927	97.008	Hay grazer	7.5	Clay loam	Reproductive	8
	Roadside	30.934	97.013	Grass	7.7	Clay loam	Reproductive	8
Thrall	Field	30.606	97.289	Corn	7.7	Clay loam	Reproductive	10
	Roadside	30.598	97.285	Grass	7.1	Clay loam	Reproductive	9
Eagle Lake	Field	29.261	95.926	Pasture	7.4	Clay loam	Reproductive	10
	Roadside	29.255	95.932	Grass	7.3	Sandy clay loam	Reproductive	9

^aReproductive stage refers to plants with mature panicles; Vegetative phase ranges from seedling to flowering initiation stage.

^bPlant density/biomass production scoring: 10 represents the highest possible density and biomass in the site.

moisture content. Ammonium and nitrate were determined by the colorimetric method (Doane & Horwath, 2003; Forster, 1995; Verdouw et al., 1978). A standard curve was developed in the spectrophotometer with the known concentrations of the standard solution, and the samples were analyzed. The absorbance recorded was then multiplied with a dilution factor and the soil moisture content to obtain the final ammonium and nitrate concentrations. The ammonium to total N ratio (ammonium concentration/[ammonium + nitrate concentration]) was then calculated.

Statistical analysis was performed using Jmp Pro16 (SAS Institute), and the results were plotted using GraphPad Prism 9. Soil collection sites were treated as the fixed effect and replications were treated as the random effect. A generalized linear model was fit to the data, which was followed by mean separation using Tukey's honestly significant difference method ($\alpha = 0.05$). To test the difference between the field and roadside johnsongrass for ammonium to total N ratio, a Student's *t*-test was conducted in Jmp.

3 | RESULTS AND DISCUSSION

3.1 | Ammonium retention in the cropland versus roadside habitats

In the majority of survey locations with paired field versus roadside comparisons (four out of five), the amount of ammonium retained by roadside johnsongrass populations

was greater (up to 40% greater) than that of the adjacent cultivated field populations (Figure 1a). When averaged across all locations within each category, the roadside johnsongrass retained much greater levels of ammonium than cultivated fields (Figure 1c). High ammonium retention in roadside johnsongrass could be a combined result of less/lack of disturbance and adaptive evolution under resource competition. Rice and Pancholy (1972) and Sylvester-Bradley et al. (1988) reported that the rate of nitrification is greater in disturbed soils (i.e., ploughed and tilled soil) than in undisturbed soils. Further, due to high resource competition in roadside environments with other surrounding vegetation, improved N-use efficiency is vital for the persistence of roadside johnsongrass, which is further necessitated by the absence of external fertilization, unlike in cultivated fields (Di & Cameron, 2002). Therefore, retaining more ammonium through inhibiting nitrification or other processes is advantageous to roadside johnsongrass and could be an adaptive strategy for its persistence in these habitats.

The exception was Mumford, wherein no difference was observed for the ammonium to total nitrate ratio between the roadside and field johnsongrass populations. This could be attributed to a frequent mowing regime at the roadside site (personal observations), potentially leading to lower levels of ammonium retention than anticipated. The roadside johnsongrass plants at this site were only 15- to 30-cm tall at the time of the survey. The Thrall and Eagle Lake locations were excluded from this analysis since they had extremely high infestations of johnsongrass (monoculture) in the field sites,

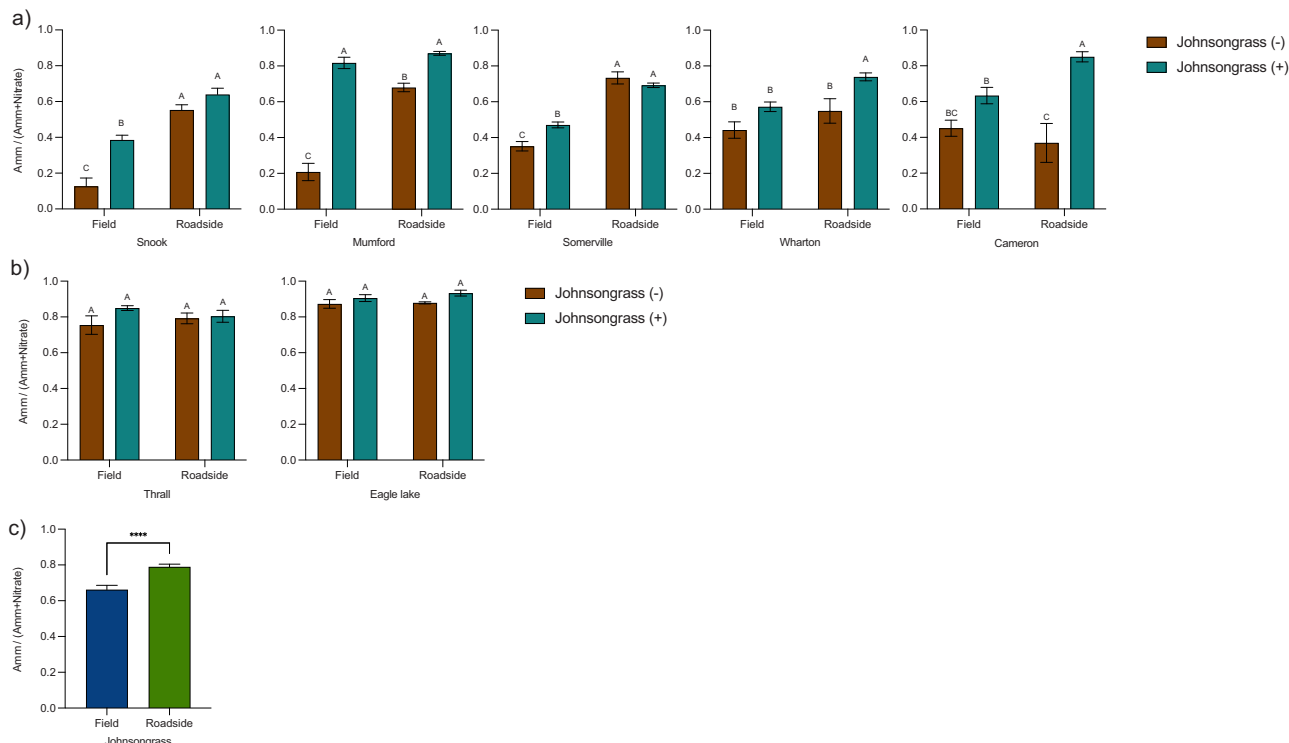


FIGURE 1 (a) The ammonium to total N ratio is plotted for different locations, each with a pair of field and roadside sites (<1 km). Bars show the mean \pm SE (standard error) of 10 plants, with three replicates each. In each location and in each site (field vs. roadside), green bars refer to the presence of johnsongrass and the brown bars indicate the absence of this species. (b) The ammonium to total N ratio is plotted for Thrall and Eagle Lake locations, which were considered a special case due to extremely high densities of johnsongrass infestations. For (a) and (b), different letters above each bar indicate significant differences, as determined by the Tukey's honestly significant difference test ($\alpha = 0.05$). (c) Average values of ammonium to total N ratio are plotted for field (blue bar) and roadside (green bar) johnsongrass for the locations mentioned in (a); a student's t -test was performed ($*p < 0.05$; $***p < 0.001$; $****p < 0.0001$) to show differences between the average values of field versus roadside johnsongrass.

with plant densities of 10 and 9 for field and roadside sites, respectively, in both locations (Table 1). Being a perennial species, it is likely that dense infestations of johnsongrass may have led to improved suppression of the nitrifying bacteria over concurrent field seasons, thus enhancing the ammonium retention levels comparable to that of roadside populations. This response, however, needs to be verified through further experimentation.

This survey was conducted as foundational research to shed light on the existence of probable nitrification-inhibition potential by johnsongrass under roadside conditions. Results suggest the likelihood of nitrification inhibition potential by johnsongrass, which, if verified, would be the first report of this ability in this species, though nitrification inhibition has been previously reported in other weed species, including wild radish, wild oats, great brome grass, and annual ryegrass (O'Sullivan et al., 2017). Results also suggest that the extent of ammonium retention by johnsongrass varies across locations, suggesting the potential influence of soil characteristics and environmental factors, which were not studied here. Soil characteristics like soil pH are reported to govern the type of BNI compound released in sorghum (Di et al., 2018). How-

ever, no correlation was found between soil pH and soil type in this study (data not shown). Further, acidic soil conditions favor ammonium retention (Lu et al., 2019; Zhang et al., 2022), but it is important to note that the soil pH is not acidic in all the survey locations (Table 1). Di et al. (2018) reported that hydrophobic exudates, such as sorgoleone, are not affected by soil pH, suggesting that the johnsongrass root exudates, if any, are likely hydrophobic. The likely nitrification inhibition ability of johnsongrass might be contributing, at least in part, to the invasiveness of this species, as has been shown in *Andropogon gayanus* (gamba grass) by Rossiter-Rachor et al. (2009), wherein this species was able to regulate soil N by decreasing nitrification.

3.2 | Soil ammonium level in the presence versus absence of johnsongrass

The ammonium to total N (ammonium + nitrate) ratio indicates the amount of ammonium retained in the plant's rhizosphere, representing the available N source. A high ammonium-to-nitrate ratio can indicate nitrification

inhibition (Subbarao & Searchinger, 2021). In general, johnsongrass is able to retain a high amount of ammonium (>60% of total N) compared to without johnsongrass, but the degree of retention varies across the locations (Figure 1a,b). Within each site (field or roadside), the johnsongrass rhizosphere soil generally had greater ammonium retention than the soil where johnsongrass was absent (Figure 1a); the exceptions include the roadside locations of Snook and Somerville, and field locations of Wharton and Cameron.

As BNI could be one of the possible explanations of high ammonium retention among other processes, the plant growth stage in the Wharton-field (very small plants at the early rhizomatous regrowth stage) and Somerville-roadside (mature plants at the peak flowering stage) may have affected the production of BNI compounds, as it is observed that BNI compound production may be greater during active vegetative growth (Aulakh et al., 2001; Subbarao et al., 2013). It is possible that plants invest more energy in flowering than exuding nitrification-inhibiting compounds (García et al., 2001). It is also likely that compounds inhibiting nitrification are synthesized and released, but are not involved in nitrification inhibition during this later stage, as BNI exudates have also been shown to have other roles in addition to nitrification inhibition (Coskun et al., 2017). In the case of small plants (Wharton-field), they may not be producing adequate amounts of root exudate to suppress nitrification. This response could also be attributed to other plant species, soil, and environmental factors (Di & Cameron, 2002; Sahrawat, 2008; Zhang et al., 2022), which were not studied here. For example, it has been shown that BNI potential in pearl millet is accelerated during drought conditions (Ghatak et al., 2022). However, there is a possibility of completely different processes in play, which needs further investigation.

Furthermore, johnsongrass in the Wharton and Cameron field locations was found in pasture field settings, with other long-term grass vegetation in the non-johnsongrass sampling sites within each location. Some of the dominant species in these pastures included broadleaf signalgrass (*Urochloa platyphylla*), Texas millet (*Urochloa texana*), and Italian ryegrass (*Lolium perenne* ssp. *multiflorum*), among others. Tropical pasture and forage grasses like *B. humidicola* (Ishikawa et al., 2003; Subbarao et al., 2007) and *L. perenne* (Wheatley et al., 1990) have been shown to have nitrification-inhibiting properties. It is possible that some pasture grasses present in the survey fields might have contributed to this, though this was not explicitly studied.

4 | CONCLUSION

Our survey results show that johnsongrass exhibits high ammonium retention in the rhizosphere, ensuring extended availability of N in the soil. The extent of ammonium

retention is greater in the case of roadside johnsongrass, suggesting the role of an undisturbed or challenging natural environment in maintaining N resources, as well as potential plant adaptation with improved N utilization. Further, the degree of ammonium retention significantly varied across the survey sites, indicating the potential influence of soil and environmental variables, for example, fertilizer application and microbiome interactions, which need further experimentation. The findings make the plausibility of johnsongrass exhibiting nitrification inhibition properties, similar to that of cultivated sorghum, as an adaptive strategy. Nevertheless, we acknowledge the possibility that other processes, such as dissimilatory nitrate reduction, leaching, and nitrous oxide emission, may be contributing to the high ammonium-to-nitrate ratio observed here, but more investigations are necessary to decipher the potential processes at play. Regardless of the mechanism, high ammonium retention in johnsongrass rhizosphere may be contributing to its adaptive success in competitive environments. Identifying and isolating this trait from johnsongrass and introgressing it into grain sorghum may be beneficial for developing climate-smart sorghum cultivars.

AUTHOR CONTRIBUTIONS

Eeshita Ghosh: Data curation; formal analysis; investigation; methodology; software; validation; visualization; writing—original draft. **Nithya Rajan:** Methodology; project administration; resources; supervision; writing—review and editing. **Dinesh Phuyal:** Methodology; resources; writing—review and editing. **Nithya Subramanian:** Methodology; project administration; resources; supervision; writing—review and editing. **Muthukumar Bagavathiannan:** Conceptualization; funding acquisition; methodology; project administration; resources; supervision; writing—review and editing.

ACKNOWLEDGMENTS

We would like to acknowledge funding support from a USDA-NIFA Foundational Program award, and field assistance from Daniel Hathcoat, Daniel Lavy, Giordano Fontana, Hayden Taylor, Ubaldo Torres, and Carson Klose.

CONFLICT OF INTEREST STATEMENT

The authors declare no conflicts of interest.

ORCID

Nithya Rajan  <https://orcid.org/0000-0003-3798-2629>

Muthukumar Bagavathiannan  <https://orcid.org/0000-0002-1107-7148>

REFERENCES

- Anderson, R., Bayer, P. E., & Edwards, D. (2020). Climate change and the need for agricultural adaptation. *Current Opinion in Plant Biology*, 56, 197–202. <https://doi.org/10.1016/j.pbi.2019.12.006>

- Aulakh, M. S., Wassmann, R., Bueno, C., Kreuzwieser, J., & Rennenberg, H. (2001). Characterization of root exudates at different growth stages of ten rice (*Oryza sativa* L.) cultivars. *Plant Biology*, 3(3), 298–298. <https://doi.org/10.1055/s-2001-15205>
- Brisson, V. L., Schmidt, J. E., Northen, T. R., Vogel, J. P., & Gaudin, A. C. M. (2019). Impacts of maize domestication and breeding on rhizosphere microbial community recruitment from a nutrient depleted agricultural soil. *Scientific Reports*, 9(1), Article 15611. <https://doi.org/10.1038/s41598-019-52148-y>
- Bulgarelli, D., Garrido-Oter, R., Münch, P. C., Weiman, A., Dröge, J., Pan, Y., McHardy, A. C., & Schulze-Lefert, P. (2015). Structure and function of the bacterial root microbiota in wild and domesticated barley. *Cell Host & Microbe*, 17(3), 392–403. <https://doi.org/10.1016/j.chom.2015.01.011>
- Coskun, D., Britto, D. T., Shi, W., & Kronzucker, H. J. (2017). Nitrogen transformations in modern agriculture and the role of biological nitrification inhibition. *Nature Plants*, 3, Article 17074. <https://doi.org/10.1038/nplants.2017.74>
- Di, H. J., & Cameron, K. C. (2002). Nitrate leaching in temperate agroecosystems: Sources, factors and mitigating strategies. *Nutrient Cycling in Agroecosystems*, 64(3), 237–256. <https://doi.org/10.1023/A:1021471531188>
- Di, T., Afzal, M. R., Yoshihashi, T., Deshpande, S., Zhu, Y., & Subbarao, G. V. (2018). Further insights into underlying mechanisms for the release of biological nitrification inhibitors from sorghum roots. *Plant and Soil*, 423(1–2), 99–110. <https://doi.org/10.1007/s11104-017-3505-5>
- Doane, T. A., & Horwath, W. R. (2003). Spectrophotometric determination of nitrate with a single reagent. *Analytical Letters*, 36(12), 2713–2722. <https://doi.org/10.1081/AL-120024647>
- EPA. (2024). *Inventory of U.S. Greenhouse Gas Emissions and Sinks: 1990–2019* (430-R-21-005). <https://www.epa.gov/ghgemissions/inventory-us-greenhouse-gas-emissions-and-sinks-1990-2019>
- Forster, J. C. (1995). 3 - Soil sampling, handling, storage and analysis. In K. Alef & P. Nannipieri (Eds.), *Methods in applied soil microbiology and biochemistry* (pp. 49–121). Academic Press. <https://doi.org/10.1016/B978-012513840-6/50018-5>
- Ghatak, A., Schindler, F., Bachmann, G., Engelmeier, D., Bajaj, P., Brenner, M., Fragner, L., Varshney, R. K., Subbarao, G. V., Chaturvedi, P., & Weckwerth, W. (2022). Root exudation of contrasting drought-stressed pearl millet genotypes conveys varying biological nitrification inhibition (BNI) activity. *Biology and Fertility of Soils*, 58(3), 291–306. <https://doi.org/10.1007/s00374-021-01578-w>
- Hakim, S., Naqqash, T., Nawaz, M. S., Laraib, I., Siddique, M. J., Zia, R., Mirza, M. S., & Imran, A. (2021). Rhizosphere engineering with plant growth-promoting microorganisms for agriculture and ecological sustainability. *Frontiers in Sustainable Food Systems*, 5, 617157. <https://doi.org/10.3389/fsufs.2021.617157>
- Hayatsu, M., Tago, K., & Saito, M. (2008). Various players in the nitrogen cycle: Diversity and functions of the microorganisms involved in nitrification and denitrification. *Soil Science and Plant Nutrition*, 54(1), 33–45. <https://doi.org/10.1111/j.1747-0765.2007.00195.x>
- Hu, H.-W., Xu, Z.-H., & He, J.-Z. (2014). Ammonia-oxidizing archaea play a predominant role in acid soil nitrification. *Advances in Agronomy*, 125, 261–302. <https://doi.org/10.1016/B978-0-12-800137-0.00006-6>
- Ishikawa, T., Subbarao, G. V., Ito, O., & Okada, K. (2003). Suppression of nitrification and nitrous oxide emission by the tropical grass *Brachiaria humidicola*. *Plant and Soil*, 255(1), 413–419. <https://doi.org/10.1023/A:1026156924755>
- Klein, P., & Smith, C. M. (2020). Invasive johnsongrass, a threat to native grasslands and agriculture. *Biologia*, 76, 413–420. <https://doi.org/10.2478/s11756-020-00625-5>
- Kowalchuk, G. A., & Stephen, J. R. (2001). Ammonia-oxidizing bacteria: A model for molecular microbial ecology. *Annual Review of Microbiology*, 55(1), 485–529. <https://doi.org/10.1146/annurev.micro.55.1.485>
- Lu, Y., Zhang, X., Jiang, J., Kronzucker, H. J., Shen, W., & Shi, W. (2019). Effects of the biological nitrification inhibitor 1,9-decanediol on nitrification and ammonia oxidizers in three agricultural soils. *Soil Biology and Biochemistry*, 129, 48–59. <https://doi.org/10.1016/j.soilbio.2018.11.008>
- Lucas García, J. A., Barbas, C., Probanza, A., Barrientos, M. L., & Gutierrez Mañero, F. J. (2001). Low molecular weight organic acids and fatty acids in root exudates of two *Lupinus* cultivars at flowering and fruiting stages. *Phytochemical Analysis*, 12(5), 305–311. <https://doi.org/10.1002/pca.596>
- Mueller, S. M., Messina, C. D., & Vyn, T. J. (2019). Simultaneous gains in grain yield and nitrogen efficiency over 70 years of maize genetic improvement. *Scientific Reports*, 9(1), Article 9095. <https://doi.org/10.1038/s41598-019-45485-5>
- Mulvaney, R. L. (1996). Nitrogen—inorganic forms. In *Methods of soil analysis: Part 3 Chemical Methods* (Vol. 5, pp. 1123–1184). SSSA. <https://doi.org/10.2136/sssabookser5.3.c38>
- Ohadi, S., Hodnett, G., Rooney, W., & Bagavathiannan, M. (2017). Gene flow and its consequences in *Sorghum* spp. *Critical Reviews in Plant Sciences*, 36, 367–385. <https://doi.org/10.1080/07352689.2018.1446813>
- O'sullivan, C. A., Whisson, K., Treble, K., Roper, M. M., Micin, S. F., & Ward, P. R. (2017). Biological nitrification inhibition by weeds: Wild radish, brome grass, wild oats and annual ryegrass decrease nitrification rates in their rhizospheres. *Crop and Pasture Science*, 68(8), 798. <https://doi.org/10.1071/CP17243>
- Rice, E. L., & Pancholy, S. K. (1972). Inhibition of nitrification by climax ecosystems. *American Journal of Botany*, 59(10), 1033–1040. <https://doi.org/10.1002/j.1537-2197.1972.tb10183.x>
- Rossiter-Rachor, N. A., Setterfield, S. A., Douglas, M. M., Hutley, L. B., Cook, G. D., & Schmidt, S. (2009). Invasive *Andropogon gayanus* (gamba grass) is an ecosystem transformer of nitrogen relations in Australian savanna. *Ecological Applications: A Publication of the Ecological Society of America*, 19(6), 1546–1560. <https://doi.org/10.1890/08-0265.1>
- Sahrawat, K. L. (2008). Factors affecting nitrification in soils. *Communications in Soil Science and Plant Analysis*, 39(9–10), 1436–1446. <https://doi.org/10.1080/00103620802004235>
- Singh, S. N., & Verma, A. (2017). Environmental review: The potential of nitrification inhibitors to manage the pollution effect of nitrogen fertilizers in agricultural and other soils: A review. *Environmental Practice*, 9(4), 266–279. <https://doi.org/10.1017/S1466046607070482>
- Soldan, R., Fusi, M., Cardinale, M., Daffonchio, D., & Preston, G. M. (2021). The effect of plant domestication on host control of the microbiota. *Communications Biology*, 4(1), Article 936. <https://doi.org/10.1038/s42003-021-02467-6>

- Stark, C. H., & Richards, K. G. (2008). The continuing challenge of agricultural nitrogen loss to the environment in the context of global change and advancing research. *Dynamic Soil, Dynamic Plant*, 2(1), 1–12.
- Subbarao, G. V., Ishikawa, T., Ito, O., Nakahara, K., Wang, H. Y., & Berry, W. L. (2006). A bioluminescence assay to detect nitrification inhibitors released from plant roots: A case study with *Brachiaria humidicola*. *Plant and Soil*, 288(1), 101–112. <https://doi.org/10.1007/s11104-006-9094-3>
- Subbarao, G. V., Nakahara, K., Ishikawa, T., Ono, H., Yoshida, M., Yoshihashi, T., Zhu, Y., Zakir, H. A. K. M., Deshpande, S. P., Hash, C. T., & Sahrawat, K. L. (2013). Biological nitrification inhibition (BNI) activity in sorghum and its characterization. *Plant and Soil*, 366(1–2), 243–259. <https://doi.org/10.1007/s11104-012-1419-9>
- Subbarao, G. V., Rondon, M., Ito, O., Ishikawa, T., Rao, I. M., Nakahara, K., Lascano, C., & Berry, W. L. (2007). Biological nitrification inhibition (BNI)—is it a widespread phenomenon? *Plant and Soil*, 294(1–2), 5–18. <https://doi.org/10.1007/s11104-006-9159-3>
- Subbarao, G. V., & Searchinger, T. D. (2021). A “more ammonium solution” to mitigate nitrogen pollution and boost crop yields. *Proceedings of the National Academy of Sciences*, 118(22), e2107576118. <https://doi.org/10.1073/pnas.2107576118>
- Subbarao, G. V., Yoshihashi, T., Worthington, M., Nakahara, K., Ando, Y., Sahrawat, K. L., Rao, I. M., Lata, J.-C., Kishii, M., & Braun, H.-J. (2015). Suppression of soil nitrification by plants. *Plant Science: An International Journal of Experimental Plant Biology*, 233, 155–164. <https://doi.org/10.1016/j.plantsci.2015.01.012>
- Sylvester-Bradley, R., Mosquera, D., & Méndez, J. E. (1988). Inhibition of nitrate accumulation in tropical grassland soils: Effect of nitrogen fertilization and soil disturbance. *Journal of Soil Science*, 39(3), 407–416. <https://doi.org/10.1111/j.1365-2389.1988.tb01226.x>
- Verdouw, H., Van Echteld, C. J. A., & Dekkers, E. M. J. (1978). Ammonia determination based on indophenol formation with sodium salicylate. *Water Research*, 12(6), 399–402. [https://doi.org/10.1016/0043-1354\(78\)90107-0](https://doi.org/10.1016/0043-1354(78)90107-0)
- Vetsch, J. A., Randall, G. W., & Fernández, F. G. (2019). Nitrate loss in subsurface drainage from a corn-soybean rotation as affected by nitrogen rate and nitrpyrin. *Journal of Environmental Quality*, 48(4), 988–994. <https://doi.org/10.2134/jeq2018.11.0415>
- Vitousek, P., & Howarth, R. (1991). Nitrogen limitation on land and in the sea: How can it occur? *Biogeochemistry*, 13(2), 87–115. <https://doi.org/10.1007/BF00002772>
- Wheatley, R., Ritz, K., & Griffiths, B. (1990). Microbial biomass and mineral N transformations in soil planted with barley, ryegrass, pea or turnip. *Plant and Soil*, 127(2), 157–167. <https://doi.org/10.1007/BF00014422>
- Zhang, M., Zeng, H., Afzal, M. R., Gao, X., Li, Y., Subbarao, G. V., & Zhu, Y. (2022). BNI-release mechanisms in plant root systems: Current status of understanding. *Biology and Fertility of Soils*, 58(3), 225–233. <https://doi.org/10.1007/s00374-021-01568-y>

How to cite this article: Ghosh, E., Rajan, N., Phuyal, D., Subramanian, N., & Bagavathiannan, M. (2024). High rhizospheric ammonium levels in *Sorghum halepense* (johnsongrass) suggests nitrification inhibition potential. *Agricultural & Environmental Letters*, 9, e20137. <https://doi.org/10.1002/acl2.20137>