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**Abstract**

Plants acclimate to increasing CO2 by reducing leaf nutrient allocation and photosynthetic capacity at the leaf level, a response that often occurs alongside growth stimulation at the whole plant level. Nutrient limitation has been hypothesized to be the primary driver of plant acclimation to CO2, as nutrient availability commonly limits primary productivity and may decrease with increasing CO2 over time. However, recent work leveraging photosynthetic least-cost theory indicates that these acclimation responses may instead be the result of optimal resource investment toward photosynthetic capacity, which maximizes nutrient allocation to whole plant growth. To understand whether nutrient limitation or optimal leaf resource investment controls plant acclimation to CO2 and how nutrient acquisition strategy modifies these responses, we grew soybean under two atmospheric CO2 levels, two inoculation treatments, and nine soil nitrogen fertilization treatments in a full factorial growth chamber experiment.

We found that …

These results suggest that XX is the dominant control of plant acclimation responses to CO2, providing important empirical data needed to refine our understanding of mechanisms driving plant acclimation to CO2.

**Keywords**

photosynthetic acclimation, soil nutrient availability, nutrient acquisition, global change

**Introduction**

Plants grown under elevated CO2 generally have less leaf nutrient content than those grown under ambient CO2 (Curtis, 1996). This reduction in leaf nutrient allocation reflects an acclimation response that corresponds with reductions in photosynthetic capacity at the leaf level and occurs alongside biomass stimulation over time at the whole plant level (Makino et al., 1997). Some have hypothesized that nutrient limitation may be the primary control of plant acclimation to CO2, as nutrient availability commonly limits primary productivity and may decrease over time in elevated CO2 environments (Fay et al., 2015; LeBauer & Treseder, 2008; Liang et al., 2016; Luo et al., 2004). The nutrient limitation hypothesis predicts that plants decrease leaf nutrient allocation and photosynthetic capacity as a direct response to progressive reductions in soil nutrient availability due to elevated CO2. The nutrient limitation hypothesis also predicts an acute stimulation in whole plant growth due to elevated CO2 that dampens over time because of progressive nutrient limitation. An alternative hypothesis to the leaf response, based on photosynthetic least-cost theory (Prentice et al., 2014; Wright et al., 2003) suggests that plants growing under elevated CO2 environments instead downregulate nutrient allocation to Rubisco to optimize resource use efficiencies at the leaf level, which maximizes resource allocation to whole plant growth. The nutrient limitation and least-cost hypotheses predict similar leaf responses to CO2, but result in different outcomes at the whole plant level.

Nutrient acquisition strategy, or the method in which plants acquire nutrients, may also impact how plants acclimate to CO2 (Smith & Keenan, 2020; Terrer et al., 2018). Plants acquire nutrients via direct uptake from their rooting systems or through symbiotic associations with mycorrhizal fungi or symbiotic nitrogen-fixing bacteria. In plants that form associations with microbial symbionts, plants allocate recent photosynthate belowground in exchange for nutrients acquired by microbial symbionts. However, not all microbial symbioses require the same belowground carbon investments to exchange nutrients. Carbon costs to acquire nitrogen, or the amount of carbon plants allocate belowground per nitrogen acquired, vary across nutrient acquisition strategies and soil nutrient availability thresholds (Perkowski et al., 2021). Interestingly, a recent global meta-analysis indicates that carbon costs to acquire nitrogen may modify plant acclimation responses to CO2 (Terrer et al., 2016, 2018), although manipulation experiments that directly test the mechanisms driving these responses are rare.

In this study, I will investigate the influence of inoculation with symbiotic nitrogen-fixing bacteria and direct soil nutrient manipulation on soybean (*Glycine max* L.) acclimation responses to CO2. This experiment will determine whether nutrient limitation or optimal leaf resource investment is the primary driver of plant acclimation to CO2 and how nutrient acquisition strategy modifies these responses. I hypothesize that leaf acclimation to CO2 will be driven by optimal leaf resource investment, not nutrient limitation. Specifically, I predict that increasing CO2 will decrease stomatal conductance, leaf nutrient allocation, and photosynthesis independent of nutrient acquisition strategy or soil nutrient availability, which will maximize resource allocation to whole plant growth. While I do not expect that soil nutrients or acquisition strategy will modify leaf acclimation responses to CO2, I do expect that soil nutrient availability will increase the positive effect of CO2 on whole plant growth. I also predict that inoculation with nitrogen-fixing bacteria will increase whole plant growth responses to CO2. However, I only expect an inoculation effect in low soil nutrient environments, as inoculated individuals should shift away from nitrogen fixation and toward direct uptake with increasing soil nutrient availability (Perkowski et al., 2021; Rastetter et al., 2001).

**Methods**

*Seed treatments and experimental design*

*Glycine max* seeds were planted in 144 6-liter pots (NS-600, Nursery Supplies, Orange, CA, USA) containing a soil-less mix containing 70% *Sphagnum* peat moss and 30% sand by volume. The soil-less mix was steam sterilized at 95C for 4 hours to eliminate any bacterial or fungal growth. Seventy-two pots were randomly selected to be planted with seeds inoculated with *Bradyrhizobium japonicum* (Verdesian N-Dure™ Soybean, Cary, NC, USA) following a brief surface sterilization in 2% sodium hypochlorite. The remaining 72 pots were planted with seeds that did not receive any inoculation treatment. Uninoculated seeds were also surface sterilized in 2% sodium hypochlorite to ensure that the only difference between seed treatments was the inoculation treatment.

Upon planting, 36 pots of each inoculation treatment were randomly placed in one of two atmospheric CO2 treatments: 420 and 1000 μmol mol-1 CO2. Pots within each unique CO2 and inoculation treatment were randomly selected to receive one of nine nitrogen fertilization treatments as a modified Hoagland’s solution (Hoagland & Arnon, 1950) equivalent to 0, 35, 70, 105, 140, 210, 280, 350, or 630 ppm N. Modified Hoagland’s solutions were designed to keep concentrations of other macronutrients and micronutrients equivalent across treatments (Table S1), and were received as 150 mL as topical agents to the soil surface of each pot twice per week.

All individuals were well watered to minimize chances of water stress. We also kept other ancillary chamber climate settings constant across treatment combinations. We simulated daytime environments using a 16-hour photoperiod, with incoming light radiation set to XX μmol m-2 s-1, temperature set to 25°C, and relative humidity set to 50%. The remaining 8 hours simulated nighttime growing conditions, with incoming light radiation set to 0 μmol m-2 s-1, temperature set to 17°C, and relative humidity again set to 50%. To better represent natural transitions between day and night conditions, we ramped temperature and incoming light radiation from daytime to nighttime or from nighttime to daytime over a two-hour period. All individuals grew under these treatment combinations and growing conditions for a six-week growth period. This treatment setup and sample size created 4 replicates per treatment combination, which power analyses suggest yield enough statistical power (β>0.8) to adequately quantify interactions between soil nutrient availability, inoculation status, and CO2.

*Leaf gas exchange*

Starting on the fourth week of the experiment, we collected weekly gas exchange measurements on the most recent fully expanded leaf. First, we surveyed net photosynthesis (*A*net; μmol m-2 s-1), stomatal conductance (*g*s; mmol mol-1), intercellular CO2 (*C*i; µmol mol-1), and chlorophyll fluorescence data. Survey gas exchange data were collected after allowing a leaf to stabilize in a cuvette where reference CO2 was set to 420 μmol mol-1, relative humidity was stabilized at 50%, cuvette temperature was set to 25°C, and incoming light radiation was set to 1500 μmol m-2 s-1.

Upon completion of survey gas exchange data, we measured CO2 response curves using the dynamic assimilation technique with a Li-COR LI-6800 portable photosynthesis machine (Li-COR Biosciences, Lincoln, Nebraska, USA). The dynamic assimilation technique eliminates the need for steady-state measurements along an atmospheric CO2 gradient, expedites CO2 response curves to a few minutes, provides >50 points to fit the response curve, and has been shown to correspond well with traditional steady-state CO2 response curves (Saathoff & Welles, 2021). We conducted dynamic CO2 response curves using the split method, which measured *A*net, *g*s, and *C*i along a ramp down from 420 µmol mol-1 CO2 to 20 µmol mol-1 CO2, followed by a ramp up from 420 µmol mol-1 CO2 to 1620 µmol mol-1 CO2 after a one-minute wait period at 420 µmol mol-1 CO2. CO2 ramps were done using the same cuvette conditions as the survey measurements explained above and were ramped at a rate of 200 μmol mol-1 CO2 min-1.

To verify that the dynamic assimilation technique matched patterns expected from traditional steady-state CO2 response curves, we randomly collected paired CO2 response curves for each week of gas exchange measurements. We targeted paired measurements for the 0 ppm N, 210 ppm N, and 630 ppm N treatments within each unique CO2 x inoculation treatment combination for a total of 12 paired response curves each week. Paired response curves were collected by first conducting the split dynamic response curve explained in the previous paragraph. The leaf was then unclamped and placed back into its growth chamber for 30 minutes to allow the leaf to return to its growing conditions. After 30 minutes, the same leaf was then reattached to the LI-6800 and allowed to stabilize to chamber conditions. We then conducted a steady state CO2 response curve to measure *A*net, *g*s, and *C*i at the following reference CO2 concentrations (*C*a; μmol mol-1): 420, 320, 220, 120, 70, 420, 420, 620, 820, 1020, 1220, and 1620. Steady state response curves were measured using the save cuvette conditions as the dynamic response curves and survey measurements. Importantly, we observed no apparent bias in steady state or dynamic assimilation response curves, confirmed both visually and through curve fitting. These results are included in the supplemental information (Table S2; Fig. S1)

Following survey and response curve measurements, we subjected individuals to at least a 30-minute period of no light and quantified dark respiration (*R*d; μmol m-2 s-1), again using a Li-COR LI-6800 with the cuvette reference CO2 set to 420 μmol mol-1, relative humidity set to 50%, cuvette temperature set to 25°C, and incoming light radiation was set to 0 μmol m-2 s-1.

*Leaf trait measurements*

On the final week of the experiment, leaf trait measurements were collected on the same focal leaf used to generate dynamic CO2 response curves. Images of each leaf were curated using a flat-bed scanner to determine wet leaf area using the 'LeafArea' R package (Katabuchi, 2015), which automates leaf area calculations using ImageJ software (Schneider et al., 2012). Each leaf was dried at 65C for at least 48 hours, and subsequently weighed and ground until homogenized. Specific leaf area (cm2 g-1) was calculated as the ratio of wet leaf area to dry leaf biomass. Using subsamples of ground and homogenized leaf biomass, we also determined leaf nitrogen content (*N*mass; g g-1) through elemental combustion analysis (Costech-4010, Costech, Inc., Valencia, CA, USA), and sent samples to the University of California-Davis Stable Isotope Facility to determine leaf δ13C and δ15N. Leaf nitrogen mass per unit leaf area (*N*area; g m-2) was calculated by dividing *N*mass by specific leaf area, then multiplying by 10,000 to convert cm-2 to m-2.

We used leaf δ13C values to estimate the ratio of intercellular (*C*i) to extracellular (*C*a) CO2 (χ; Pa Pa-1) following the approach of Farquhar *et al.* (1989) described in Cernusak *et al.* (2013). While intercellular and extracellular CO2 concentrations were directly measured during each CO2 response curve, deriving χ from δ13C provides a more integrative estimate of the *C*i:*C*a over an individual leaf’s lifespan . We derived χ as:

(Eqn. 1)

Δ13C represents the relative difference between leaf δ13C (‰) and air δ13C (‰), and is calculated from the following equation:

(Eqn. 2)

where δ13Cair is assumed to be -8‰ (Farquhar et al., 1989; Keeling et al., 1979), *a* represents the fractionation between 12C and 13C due to diffusion in air, assumed to be 4.4‰, and *b* represents the fractionation caused by Rubisco carboxylation, assumed to be 27‰ (Farquhar et al., 1989).

*A*/*C*i *curve-fitting and parameter estimation*

We fit *A*net/*C*i curves of each individual using a custom-built function in R that estimates the maximum rate of Rubisco carboxylation (*V*cmax; µmol m-2 s-1) and maximum rate of electron transport for RuBP regeneration (*J*max; µmol m-2 s-1) based on equations in the Farquhar, von Caemmerer, and Berry biochemical model of C3 photosynthesis (Farquhar et al., 1980). For each curve fit, we removed points along the curve that we visually inferred as TPU limited points. Kinetic parameters and CO2 compensation points were estimated using leaf temperature and equations derived in Bernacchi et al. (2001) and Medlyn et al. (2002). We also included dark respiration survey measurements in our curve fits, which optimized Rubisco-limited photosynthesis and electron transport limited photosynthesis fits. Because dark respiration measurements were conducted at different leaf temperatures due to reduced incoming light radiation, we standardized dark respiration measurements to the average temperature of each respective dynamic CO2 response curve following the using a log-polynomial approach explained in Heskel *et al.* (2016), where:

(Eqn. 3)

*R*T is the temperature standardized respiration rate, *T* is the temperature in which a given respiration rate is being standardized, and *T*ref is the temperature of the respiration measurement *R*Tref. *b* and *c* are coefficients that Heskel *et al.* (2016) derived from a log-polynomial approach described in O’Sullivan *et al.* (2013) for plant functional types and biomes. We used coefficients set by Heskel *et al.* (2016) for C3 herbaceous species, where *b* was set to 0.1271 and *c* was set to -0.00110.

We then manually standardized *V*cmax and *J*max to25C using a modified Arrhenius equation (as in Kattge & Knorr, 2007):

(Eqn. 4)

*k*25 represents the standardized *V*cmax or *J*max rate at 25C, *k*obs represents the *V*cmax or *J*max estimate at the average leaf temperature measured inside the cuvette during the CO2 response curve. *H*a is the activation energy of *V*cmax (71,513 J mol-1; Kattge & Knorr, 2007) or *J*max (49,884 J mol-1; Kattge & Knorr, 2007). *H*d represents the deactivation energy of both *V*cmax and *J*max (200,000 J mol-1; Medlyn et al., 2002), and R represents the universal gas constant (8.314 J mol-1 K-1). *T*ref represents the standardized temperature of 298.15 K (25C) and *T*obs represents the mean leaf temperature (in K) during each CO2 response curve. ΔS is an entropy term that Kattge & Knorr (2007) derived as a linear relationship with average growing season temperature (*T*g; °C), where:

(Eqn. 5a)

and:

(Eqn. 5b)

We estimated *T*g in equations 2 and 3 based on mean daily (24-hour) air temperature of the 30 days leading up to the day of each sample collection. Temperature data were collected from a nearby weather station located on the Cornell University campus (42.449 N, 76.449 W), which was located within a 20-km radius of all sites. We then used *V*cmax25 and *J*max25 estimates to calculate the ratio of *J*max25 to *V*cmax25 (*J*max25:*V*cmax25; unitless).

Finally, we standardized dark respiration to 25C (*R*d25; μmol m-2 s-1) using the log-polynomial explained in Eq. XX and the same coefficients explained above, with *T*ref set to 25C.

*Stomatal limitation*

We quantified the extent by which stomatal conductance limited photosynthesis (*l*; unitless) following equations originally described in Farquhar & Sharkey (1982). Stomatal limitation is an index where values that approach 1 indicate that net photosynthesis is becoming more limited by stomatal conductance, and is calculated as:

(Eqn. 6)

*A*net represents the net photosynthesis rate measured at 400 μmol mol-1 CO2, while *A*mod represents the photosynthetic rate where *C*i = *C*a. *A*mod was calculated as:

(Eqn. 7)

*V*cmax represents the temperature unstandardized maximum rate of Rubisco carboxylation. We used the temperature unstandardized *V*cmax value because *A*net values were not standardized to 25°C. *R*d represents dark respiration, which was the dark respiration value we temperature standardized to the CO2 response curve fit. Γ\* (Pa) is the CO2 compensation point in the absence of dark respiration, while *K*m is the Michaelis-Menten coefficient for Rubisco-limited photosynthesis. *K*m was calculated as:

(Eqn. 8)

*K*c refers to the Michaelis-Menten coefficient for Rubisco affinity to CO2, *K*o refers to the Michaelis-Menten coefficient for Rubisco affinity to O2, and *O*i refers to leaf intercellular O2 concentrations. Γ\* and *K*m were standardized to the average temperature of each CO2 response curve using equations and parameters described in Bernacchi et al. (2001).

*Tradeoffs between nitrogen and water use*

Photosynthetic nitrogen use efficiency (*PNUE*; µmol CO2 gN-1 s-1) was calculated by dividing *A*net measured at 420 μmol mol-1 CO2 by *N*area. We used χ, mentioned above, to estimate water use efficiency. Tradeoffs between nitrogen and water use were determined by calculating the ratio of *N*area to *g*s measured at 420 μmol mol-1 CO2 (*N*area: *g*s; gN s mol-1 H2O) and *V*cmax to *g*s measured at 420 μmol mol-1 CO2 (*V*cmax: *g*s; μmol CO2 mol-1 H2O), as done in Paillassa et al. (2020) and Bialic‐Murphy et al. (2021). We used the temperature unstandardized *V*cmax value instead of *V*cmax25 for *V*cmax: *g*s because stomatal conductance values were not standardized to 25°C.

*Whole plant traits*

The day after dynamic CO2 response curves were collected on the sixth week, we harvested all experimental individuals and separated biomass of each experimental individual into major organ types (leaves, stems, roots, and root nodules when present) approximately seven weeks after experiment initiation. Leaf areas of all harvested leaves were measured using an LI-3100C (Li-COR Biosciences, Lincoln, Nebraska, USA). Total leaf area (cm2) was calculated as the sum of all leaf areas, and included the focal leaf measured during the dynamic CO2 response curve. All harvested material was dried in an oven set to 65°C for at least 48 hours, weighed, and ground to homogeneity. Total dry biomass (g) was calculated as the sum of dry leaf, stem, root, and root nodule biomass. We also quantified carbon and nitrogen content through elemental combustion (Costech-4010, Costech, Inc., Valencia, CA, USA) of each respective organ type using subsamples of ground and homogenized organ tissue.

Following the approach explained in Perkowski et al. (2021), we calculated structural carbon costs to acquire nitrogen as the ratio of total belowground carbon biomass to whole plant nitrogen biomass (g C g-1 N). Belowground carbon biomass (g C) was calculated by multiplying the carbon content of roots and root nodules by total biomass of each respective organ type, then adding root carbon biomass and root nodule carbon biomass. Whole plant nitrogen biomass (g N) was calculated by multiplying the nitrogen content of leaves, stems, roots, and root nodules by biomass of each respective organ type, then calculating the sum of nitrogen biomass of each organ type. This calculation only quantifies plant structural carbon costs to acquire nitrogen and does not include any additional carbon costs of nitrogen acquisition that are associated with root respiration, root exudation, or root turnover. An explicit explanation of the limitations for interpreting this calculation can be found in Perkowski et al. (2021) and Terrer et al. (2018).

*Nitrogen fixation*

We calculated plant investments in nitrogen fixation as the ratio of root nodule biomass to root biomass, where increasing values indicate an increase in plant investments to nitrogen fixation (Dovrat et al., 2018, 2020; Perkowski et al., 2021). We also calculated the percent of leaf nitrogen acquired from the atmosphere (Nfda; unitless) using leaf δ15N and the following equation from Andrews et al. (2011):

(Eqn. 9)

where δ15Nreference refers to a reference plant that exclusively acquires nutrients via direct uptake, δ15Nsample refers to an individual’s leaf δ15N, and B refers to individuals that are entirely reliant on nitrogen fixation. Within each nitrogen fertilization treatment x CO2 treatment combination (n=18), we calculated the mean leaf δ15N for individuals growing in the non-inoculated treatment for δ15Nreference. Any individuals with visual confirmation of root nodule formation or nodule initiation were omitted from the calculation of δ15Nreference. Following recommendations from Andrews et al. (2011) we calculated B within each CO2 treatment by calculating the mean leaf δ15N of inoculated individuals that formed nodules. We did not calculate B within each unique soil nitrogen x CO2 treatment combination, as previous studies suggest decreased reliance on nitrogen fixation with increasing soil nitrogen availability (Perkowski et al., 2021). This approach for estimating nitrogen fixation standardizes values such that approaching 1 indicates increasing reliance on nitrogen fixation, while values that approach 0 indicate decreasing reliance on nitrogen fixation.

*Statistical analyses*

We built a series of linear mixed-effects models to investigate the impacts of atmospheric CO2, soil nitrogen fertilization, and inoculation with *B. japonicum* on *G. max* leaf photosynthesis, tradeoffs between nitrogen and water use, whole plant growth, and reliance on nitrogen fixation. All models included atmospheric CO2 and inoculation treatments as categorical fixed effects, and soil nitrogen fertilization as an additional continuous fixed effect. Models also included interaction terms between all three fixed effects. Growth chamber letter within each CO2 treatment (A-C) and growth chamber number (1-6) were included as additional random intercept terms to account for any microclimate differences between growth chambers within and across CO2 treatments. Models with this independent structure were created for each of the following dependent variables: *N*area, *SLA*, *N*mass, *A*net, *V*cmax25, *J*max25, *J*max25:*V*cmax25, *R*d25, *R*d25:*V*cmax25,total leaf area, whole plant biomass, *g*s, χ, *PNUE*, *iWUE*, *N*area:*g*s, *V*cmax:*g*s, structural carbon costs to acquire nitrogen, belowground carbon biomass, whole plant nitrogen biomass, total biomass, total leaf area, root nodule biomass: root biomass, root nodule biomass, root biomass, and %N from the atmosphere.

We used Shapiro-Wilk tests of normality to determine whether linear mixed-effects models satisfied residual normality assumptions. All models satisfied residual normality assumptions except [add traits here] (Shapiro-Wilk: p<0.05 in all cases). We attempted to satisfy residual normality assumptions for these dependent variables by first fitting models using dependent variables that were natural log transformed. If residual normality assumptions were still not met after a natural-log transformation (Shapiro-Wilk: p<0.05), then models were fit using dependent variables that were square root transformed. All residual normality assumptions were met with either a natural log or square root data transformation (Shapiro-Wilk: p>0.05 in all cases). Specifically, we natural log transformed[add traits here] and square root transformed [add traits here].

In all statistical models, we used the 'lmer' function in the 'lme4' R package (Bates et al., 2015) to fit each model and the 'Anova' function in the 'car' R package (Fox & Weisberg, 2019) to calculate Type II Wald's χ2 and determine the significance (α=0.05) of each fixed effect coefficient. We then used the 'emmeans' R package (Lenth, 2019) to conduct post-hoc comparisons using Tukey's tests, where degrees of freedom were approximated using the Kenward-Roger approach (Kenward & Roger, 1997). All analyses and plots were conducted in R version 4.2.0 (R Core Team, 2021).

**References**

Andrews, M., James, E. K., Sprent, J. I., Boddey, R. M., Gross, E., & dos Reis, F. B. (2011). Nitrogen fixation in legumes and actinorhizal plants in natural ecosystems: Values obtained using 15N natural abundance. *Plant Ecology and Diversity*, *4*(2–3), 117–130. https://doi.org/10.1080/17550874.2011.644343

Bernacchi, C. J., Singsaas, E. L., Pimentel, C., Portis, A. R., & Long, S. P. (2001). Improved temperature response functions for models of Rubisco-limited photosynthesis. *Plant, Cell and Environment*, *24*(2), 253–259. https://doi.org/10.1046/j.1365-3040.2001.00668.x

Bialic‐Murphy, L., Smith, N. G., Voothuluru, P., McElderry, R. M., Roche, M. D., Cassidy, S. T., Kivlin, S. N., & Kalisz, S. (2021). Invasion‐induced root–fungal disruptions alter plant water and nitrogen economies. *Ecology Letters*, *24*(6), 1145–1156. https://doi.org/10.1111/ele.13724

Cernusak, L. A., Ubierna, N., Winter, K., Holtum, J. A. M., Marshall, J. D., & Farquhar, G. D. (2013). Environmental and physiological determinants of carbon isotope discrimination in terrestrial plants. *New Phytologist*, *200*(4), 950–965. https://doi.org/10.1111/nph.12423

Curtis, P. S. (1996). A meta-analysis of leaf gas exchange and nitrogen in trees grown under elevated carbon dioxide. *Plant, Cell and Environment*, *19*(2), 127–137. https://doi.org/10.1111/j.1365-3040.1996.tb00234.x

Dovrat, G., Bakhshian, H., Masci, T., & Sheffer, E. (2020). The nitrogen economic spectrum of legume stoichiometry and fixation strategy. *New Phytologist*, *227*(2), 365–375. https://doi.org/10.1111/nph.16543

Dovrat, G., Masci, T., Bakhshian, H., Mayzlish Gati, E., Golan, S., & Sheffer, E. (2018). Drought-adapted plants dramatically downregulate dinitrogen fixation: Evidences from Mediterranean legume shrubs. *Journal of Ecology*, *106*(4), 1534–1544. https://doi.org/10.1111/1365-2745.12940

Farquhar, G. D., Ehleringer, J. R., & Hubick, K. T. (1989). Carbon isotope discrimination and photosynthesis. *Annual Review of Plant Physiology and Plant Molecular Biology*, *40*(1), 503–537. https://doi.org/10.1146/annurev.pp.40.060189.002443

Farquhar, G. D., & Sharkey, T. D. (1982). Stomatal conductance and photosynthesis. *Annual Review of Plant Physiology*, *33*(1), 317–345. https://doi.org/10.1146/annurev.pp.33.060182.001533

Farquhar, G. D., von Caemmerer, S., & Berry, J. A. (1980). A biochemical model of photosynthetic CO*2* assimilation in leaves of C3 species. *Planta*, *149*(1), 78–90. https://doi.org/10.1007/BF00386231

Fay, P. A., Prober, S. M., Harpole, W. S., Knops, J. M. H., Bakker, J. D., Borer, E. T., Lind, E. M., MacDougall, A. S., Seabloom, E. W., Wragg, P. D., Adler, P. B., Blumenthal, D. M., Buckley, Y. M., Chu, C., Cleland, E. E., Collins, S. L., Davies, K. F., Du, G., Feng, X., … Yang, L. H. (2015). Grassland productivity limited by multiple nutrients. *Nature Plants*, *1*(7), 15080. https://doi.org/10.1038/nplants.2015.80

Heskel, M. A., O’Sullivan, O. S., Reich, P. B., Tjoelker, M. G., Weerasinghe, K. W. L. K., Penillard, A., Egerton, J. J. G., Creek, D., Bloomfield, K. J., Xiang, J., Sinca, F., Stangl, Z. R., Martinez-de la Torre, A., Griffin, K. L., Huntingford, C., Hurry, V., Meir, P., Turnbull, M. H., & Atkin, O. K. (2016). Convergence in the temperature response of leaf respiration across biomes and plant functional types. *Proceedings of the National Academy of Sciences*, *113*(14), 3832–3837. https://doi.org/10.1073/pnas.1520282113

Hoagland, D. R., & Arnon, D. I. (1950). The water-culture method for growing plants without soil. *California Agricultural Experiment Station: 347*, *347*(2), 1–32.

Katabuchi, M. (2015). LeafArea: An R package for rapid digital analysis of leaf area. *Ecological Research*, *30*(6), 1073–1077.

Kattge, J., & Knorr, W. (2007). Temperature acclimation in a biochemical model of photosynthesis: a reanalysis of data from 36 species. *Plant, Cell & Environment*, *30*(9), 1176–1190. https://doi.org/10.1111/j.1365-3040.2007.01690.x

Keeling, C. D., Mook, W. G., & Tans, P. P. (1979). Recent trends in the 13C/12C ratio of atmospheric carbon dioxide. *Nature*, *277*(5692), 121–123. https://doi.org/10.1038/277121a0

LeBauer, D. S., & Treseder, K. (2008). Nitrogen limitation of net primary productivity. *Ecology*, *89*(2), 371–379. https://doi.org/10.1890/06-2057.1

Liang, J., Qi, X., Souza, L., & Luo, Y. (2016). Processes regulating progressive nitrogen limitation under elevated carbon dioxide: a meta-analysis. *Biogeosciences*, *13*(9), 2689–2699. https://doi.org/10.5194/bg-13-2689-2016

Luo, Y., Currie, W. S., Dukes, J. S., Finzi, A. C., Hartwig, U. A., Hungate, B. A., McMurtrie, R. E., Oren, R., Parton, W. J., Pataki, D. E., Shaw, R. M., Zak, D. R., & Field, C. B. (2004). Progressive nitrogen limitation of ecosystem responses to rising atmospheric carbon dioxide. *BioScience*, *54*(8), 731–739. https://doi.org/10.1641/0006-3568(2004)054[0731:PNLOER]2.0.CO;2

Makino, A., Harada, M., Sato, T., Nakano, H., & Mae, T. (1997). Growth and N Allocation in Rice Plants under CO2 Enrichment. *Plant Physiology*, *115*(1), 199–203. https://doi.org/10.1104/pp.115.1.199

Medlyn, B. E., Dreyer, E., Ellsworth, D. S., Forstreuter, M., Harley, P. C., Kirschbaum, M. U. F., Le Roux, X., Montpied, P., Strassemeyer, J., Walcroft, A., Wang, K., & Loustau, D. (2002). Temperature response of parameters of a biochemically based model of photosynthesis. II. A review of experimental data. *Plant, Cell & Environment*, *25*(9), 1167–1179. https://doi.org/10.1046/j.1365-3040.2002.00891.x

O’Sullivan, O. S., Weerasinghe, K. W. L. K., Evans, J. R., Egerton, J. J. G., Tjoelker, M. G., & Atkin, O. K. (2013). High-resolution temperature responses of leaf respiration in snow gum (*Eucalyptus pauciflora*) reveal high-temperature limits to respiratory function. *Plant, Cell & Environment*, *36*(7), 1268–1284. https://doi.org/10.1111/pce.12057

Paillassa, J., Wright, I. J., Prentice, I. C., Pepin, S., Smith, N. G., Ethier, G., Westerband, A. C., Lamarque, L. J., Wang, H., Cornwell, W. K., & Maire, V. (2020). When and where soil is important to modify the carbon and water economy of leaves. *New Phytologist*, *228*(1), 121–135. https://doi.org/10.1111/nph.16702

Perkowski, E. A., Waring, E. F., & Smith, N. G. (2021). Root mass carbon costs to acquire nitrogen are determined by nitrogen and light availability in two species with different nitrogen acquisition strategies. *Journal of Experimental Botany*, *72*(15), 5766–5776. https://doi.org/10.1093/jxb/erab253

Prentice, I. C., Dong, N., Gleason, S. M., Maire, V., & Wright, I. J. (2014). Balancing the costs of carbon gain and water transport: testing a new theoretical framework for plant functional ecology. *Ecology Letters*, *17*(1), 82–91. https://doi.org/10.1111/ele.12211

Prentice, I. C., Liang, X., Medlyn, B. E., & Wang, Y.-P. (2015). Reliable, robust and realistic: The three R’s of next-generation land-surface modelling. *Atmospheric Chemistry and Physics*, *15*, 5987–6005. https://doi.org/10.5194/acp-15-5987-2015

Rastetter, E. B., Vitousek, P. M., Field, C. B., Shaver, G. R., Herbert, D., & Ågren, G. I. (2001). Resource optimization and symbiotic nitrogen fixation. *Ecosystems*, *4*(4), 369–388. https://doi.org/10.1007/s10021-001-0018-z

Saathoff, A. J., & Welles, J. (2021). Gas exchange measurements in the unsteady state. *Plant Cell and Environment*, *44*(11), 3509–3523. https://doi.org/10.1111/pce.14178

Schneider, C. A., Rasband, W. S., & Eliceiri, K. W. (2012). NIH Image to ImageJ: 25 years of image analysis. *Nature Methods*, *9*(7), 671–675. https://doi.org/10.1038/nmeth.2089

Smith, N. G., & Keenan, T. F. (2020). Mechanisms underlying leaf photosynthetic acclimation to warming and elevated CO2 as inferred from least‐cost optimality theory. *Global Change Biology*, *26*(9), 5202–5216. https://doi.org/10.1111/gcb.15212

Terrer, C., Vicca, S., Hungate, B. A., Phillips, R. P., & Prentice, I. C. (2016). Mycorrhizal association as a primary control of the CO2 fertilization effect. *Science*, *353*(6294), 72–74. https://doi.org/10.1126/science.aaf4610

Terrer, C., Vicca, S., Stöcker, B. D., Hungate, B. A., Phillips, R. P., Reich, P. B., Finzi, A. C., & Prentice, I. C. (2018). Ecosystem responses to elevated CO2 governed by plant–soil interactions and the cost of nitrogen acquisition. *New Phytologist*, *217*(2), 507–522. https://doi.org/10.1111/nph.14872

Wright, I. J., Reich, P. B., & Westoby, M. (2003). Least-cost input mixtures of water and nitrogen for photosynthesis. *The American Naturalist*, *161*(1), 98–111. https://doi.org/0003-0147/2003/16101-010387