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Direct uptake of organically derived carbon by grass roots and allocation in leaves and phytoliths: ^{13}C labeling evidence

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Abstract. In the rhizosphere, the uptake of low-molecular-weight carbon (C) and nitrogen (N) by plant roots has been well documented. While organic N uptake relative to total uptake is important, organic C uptake is supposed to be low relative to the plant's C budget. Recently, radiocarbon analyses demonstrated that a fraction of C from the soil was occluded in amorphous silica micrometric particles that precipitate in plant cells (phytoliths). Here, we investigated whether and to what extent organically derived C absorbed by grass roots can feed the C occluded in phytoliths. For this purpose we added ^{13}C- and ^{15}N-labeled amino acids (AAs) to the silicon-rich hydroponic solution of the grass *Festuca arundinacea*. The experiment was designed to prevent C leakage from the labeled nutritive solution to the chamber atmosphere. After 14 days of growth, the ^{13}C and ^{15}N enrichments (^{13}C excess and ^{15}N excess) in the roots, stems and leaves as well as phytoliths were measured relative to a control experiment in which no labeled AAs were added. Additionally, the ^{13}C excess was measured at the molecular level, in AAs extracted from roots and stems and leaves. The net uptake of labeled AA-derived ^{13}C reached 4.5 % of the total AA ^{13}C supply. The amount of AA-derived ^{13}C fi ed in the plant was minor but not nil (0.28 and 0.10 % of total C in roots and stems/leaves, respectively). Phenylalanine and methionine that were supplied in high amounts to the nutritive solution were more ^{13}C-enriched than other AAs in the plant. This strongly suggested that part of AA-derived ^{13}C was absorbed and translocated into the plant in its original AA form. In phytoliths, AA-derived ^{13}C was detected. Its concentration was on the same order of magnitude as in bulk stems and leaves (0.15 % of the phytolith C). This find ing strengthens the body of evidences showing that part of organic compounds occluded in phytoliths can be fed by C entering the plant through the roots. Although this experiment was done in nutrient solution and its relevance for soil C uptake assessment is therefore limited, we discuss plausible forms of AA-derived ^{13}C absorbed and translocated in the plant and eventually fi ed in phytoliths, and implications of our results for our understanding of the C cycle at the soil–plant–atmosphere interface

1 Introduction

In the rhizosphere, there are numerous known interactions between carbon (C) and nitrogen (N) processes that have yet to be accurately assessed in qualitative and quantitative terms for their consideration in carbon cycle models (Heimann and Reichstein, 2008). Among those interactions the uptake of low-molecular-weight C and N (e.g., organic acids, sugars and amino acids (AAs)) by plant roots (both mycorrhizal and non-mycorrhizal plants) has been well documented through labeling experiments using hydroponic solutions, artificia substrats or soils (e.g., Bardgett et al., 2003; Kuzyakov and Jones, 2006; Biernath et al., 2008; Jones et al., 2009a; Näsholm et al., 2009; Sauheitl et al., 2009; Ras-

mussen et al., 2010; Gioseff et al., 2012; Moran-Zuloaga et al., 2015). The aim of most of these studies was to investigate to what extent and under which conditions organic N could be utilized by plants as a direct source of N (i.e., without going through a mineralization step). The answers are still debated (Jones and Darrah, 1992; Jones et al., 2009a; Rasmussen et al., 2010; Moran-Zuloaga et al., 2015). In particular, evidence that plant roots can uptake labeled inorganic C, which may bias the results from organic N uptake studies when using bulk measurement of dual-labeling (^{13}C and ^{15}N), has been put forward (Biernath et al., 2008; Rasmussen et al., 2010). However, the use of molecular and position-specifi labeling technics can evidence the uptake and fixa tion of intact AAs (Sauheitl et al., 2009; Moran-Zuloaga et al., 2015). Organic C uptake was also investigated through the estimation of the net uptake of glucose C. This uptake has been shown to be low relative to the plant's C budget and has often been interpreted as the recapture of roots exudates (Jones and Darrah, 1992, 1993, 1996; Kuzyakov and Jones, 2006; Jones et al., 2009a). However, very recently, in the frame of a non-labeling experiment, radiocarbon analyses demonstrated that a fraction of C occluded in amorphous silica micrometric particles that precipitate in plant cells (phytoliths) came from old soil C (Santos et al., 2012; Reyerson et al., 2015). Silicon (Si) is the second-most-abundant element of the earth surface after oxygen. Its uptake by plants is widespread and generates, at the ecosystem scale, important flu es from the soil to plants (Conley, 2002). For instance, Si absorption represents 2 to 10 times the amount of dissolved Si exported to stream water in tropical ecosystems (Alexandre et al., 2011). If part of the soil C uptake is linked to Si uptake in the rhizosphere, the involved flu may thus also be significant

Here, we aim to investigate whether and to what extent C derived from organic forms such as AAs can be absorbed by grass roots, fi ed in the plant and ultimately feed the organic C occluded in phytoliths (phytC). We choose to focus on AAs as they are ubiquitous in soil organic matters of various residence times (Bol et al., 2009). For this purpose we added ^{13}C- and ^{15}N-labeled AAs to the Si-rich nutrient solution of the grass *Festuca arundinacea*. After 2 weeks of growth, the ^{13}C and ^{15}N enrichments in the roots, stems/leaves and phytoliths of the grass (two replicates) were quantified Enrichments were also measured at the molecular level (four replicates), in AAs of roots and stems/leaves. PhytC could not be analyzed at the molecular level, due to its very small concentration. The experiment was designed to prevent C leakage from the labeled nutritive solution to the chamber atmosphere.

2 Material and methods

2.1 Hydroponic culture

Festuca arundinacea, commonly referred to as tall fescue, is widely distributed globally as a forage and an invasive grass species (Gibson and Newman, 2001) and can adapt to a wide range of conditions. *Festuca arundinacea* was grown in hydroponic conditions for 24 days using an experimental procedure adapted from RHIZOtest (Bravin et al., 2010), a plant-based test recently standardized (ISO 16198:2015). Seventy-two plant-receiving pots (i.e., a cylinder closed at the bottom with a polyamide mesh of 30 µm pore size, using an adjustable clamp) were inserted in three perforated platforms covering three 12 L tanks containing the nutrient solutions (24 plant pots per tank) (Fig. 1). This assembly enabled close contact between seeds or seedling roots and the nutrient solutions. In order to prevent escape of the C- and N-bearing gas from the nutrient solution to the chamber atmosphere, O-rings sealed the plant pots to the perforated platform and the perforated platform to the tank. Each tank was hermetically connected to two 20 L containers (an input container fille with the nutrient solutions and a waste container). Additionally, after 10 days of germination, agar-agar (polysaccharide agarose) was added around the sprouts when they were 8 cm in height. At the same time, the germination solution was replaced by the growth solution labeled in ^{13}C and ^{15}N. The growth solution was entirely renewed once, after 8 days of growth. Otherwise the germination and nutritive solutions were renewed at a rate of 2 L 24 h^{-1} using a peristaltic pump (Fig. 1).

Overall, 10 L of germination solution and 64 L of growth solution were used per tank. Germination and the growth nutrient solution composition were described in detail in Guigues et al. (2014). The nutrient solutions included 42 of inorganic N (KNO_3) and 18 mg L^{-1} of inorganic C in ethylenediaminetetraacetic acid (EDTA), added to chelate metals the plant uses for growth. The solutions were also supplemented with 105 mg L^{-1} of SiO_2 (in the form of SiO_2K_2O). The growth chamber parameters were set at (day/night) 25/20 °C, 75/70 % relative humidity and 16/8 h with a photon flu density of 450 µmol photons m^{-2} s^{-1} during the day.

At the end of the experiment, all samples were cleaned (to remove agar-agar), rinsed in deionized water and oven-dried at 50 °C (to constant mass). When the tanks were fille (1st and 8th days of growth) and emptied (8th and 14th days of growth), the growth solution was sampled and kept frozen for future analyses.

2.2 Isotope labeling

In the two firs tanks (two replicates), a mixture containing four ^{13}C- and ^{15}N-labeled AAs was added to the growth solution containing only inorganic C and N (as described

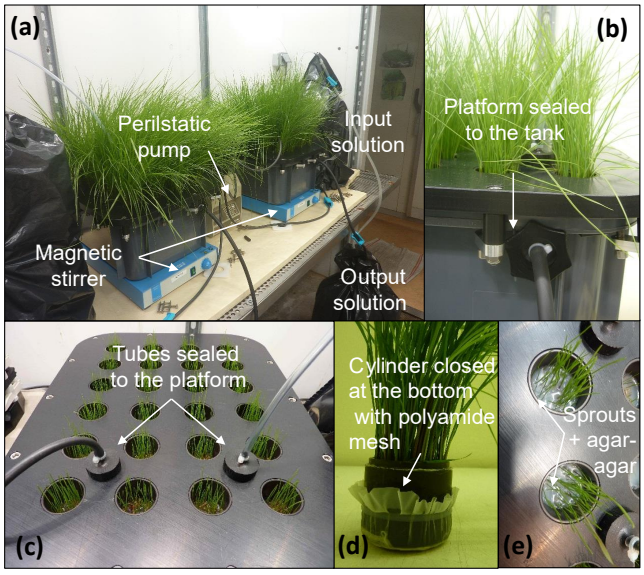

Figure 1. The labeling experiment in the growth chamber. **(a)** The two labeled tanks are connected to the solution containers. A peristaltic pump facilitates the solution renewals. **(b)** A platform is sealed to each tank. **(c)** Plant-receiving pots are cylinders closed at the bottom with a polyamide mesh. **(d)** Twenty-four plant-receiving pots are inserted into each platform. **(e)** In each pot, agar-agar was added around the sprouts to limit gas exchanges.

above). Those AAs were selected for their following characteristics: L-alanine (l-ALA) (C / N = 2.6) is ubiquist and occurs in high proportions in soils and plant proteins. The D-enantiomer of alanine (D-ALA), which is present in natural soils (Hill et al., 2011), was expected to be more resistant to degradation and, if absorbed, less subject to metabolization. Consequently, we speculated that D-ALA may accumulate as a waste product firs in cell vacuoles and later in phytoliths. L-Phenylalanine (L-PHE) (C / N = 7.7) comprises a phenolic ring resistant to decomposition by microorganisms in soils, solutions or plants. L-Methionine (L-METH) (C / N = 4.3) is a sulfur amino acid expected to be recovered at low abundance in plants, but it is easily identifi able in gas chromatography–isotope ratio mass spectrometry (GC-IRMS). Commercial 97–99 % ^{13}C and ^{15}N molecules (Euriso-top) were diluted with non-labeled amino acids to reach the following atom abundances: L-PHE (19.51 %^{13}C; 19.13 %^{15}N); L-METH (19.87 %^{13}C; 19.49 % ^{15}N); L-ALA (22.05 %^{13}C; 16.26 %^{15}N); and D-ALA (7.43 %^{13}C; 0.37 % ^{15}N). All AAs were uniformly labeled except (i) D-ALA, which was not ^{15}N-labeled but was ^{13}C-labeled on one atom (C-2), and (ii) an equivalent fraction of L-ALA because the labeled D-ALA was provided as a racemic mixture (DL-ALA). In each tank, the following amounts of AAs were added to the growth solution: PHE 249.9; MET 125.1; L-ALA 150.3; and D-ALA 125.2 mg. The total mixture (two tanks) represented 322 mg of C (26.8 mmol) and 75.7 mg of N (5.4 mmol), with average atom abundances of 18.15 % ^{13}C and 13.43 % ^{15}N. The maximum AA concentration in the growth solution on the 1st or 8th day of the growth period was 0.225 mmol L^{-1}, equivalent to 6.7 of ^{13}C and 1.6 mg L^{-1} of ^{15}N.

The third tank was only fille with the growth solution, without labeled AAs. It served as a control experiment to calculate the ^{13}C and ^{15}N enrichments of the plants from the labeled tanks and to verify that ^{13}C and ^{15}N derived from AAs (AA-derived ^{13}C and AA-derived ^{15}N), which may have contaminated the chamber CO_2, were not photosynthesized by the plants.

2.3 Analyses

For each tank (one control tank and two labeled tanks) stems/leaves were separated from the roots into two samples. The six resulting samples were ground fine than 200 µm. After alkaline fusion, they were analyzed in SiO_2 using inductively coupled plasma optical emission spectrometry (ICP-OES).

For each sample, total C, ^{13}C / ^{12}C, total N and ^{15}N / ^{14}N were determined after dry combustion by IRMS using a Carlo Erba NA 1500 elemental analyzer (EA) coupled to a Thermo-Finnigan Delta Plus mass spectrometer. Solutions were also analyzed for total C and ^{13}C / ^{12}C by IRMS, after evaporation by dry combustion in tin capsules.

Proteic carbon was analyzed as the sum of 19 individual AAs representing ca. 95 % of all AAs. Quantificatio and ^{13}C / ^{12}C determination of individual AAs were performed using a GC-IRMS (Thermo Fisher Scientific) The extraction and purificatio procedure was a slightly modifie version (Rubino et al., 2014) of the protocol developed by Amelung et al. (2006). Briefl , dry plant samples were hydrolyzed in 6 M HCl (20 h, 100 °C). AAs were purifie on Dowex 50 W X8 cation exchange resin (100–200 mesh size, Arcos Organics, Thermo Fisher Scientific) dried by rotary evaporation, re-dissolved in 0.1 M HCl and dried again by speed-vacuum evaporation. AAs were separated and quantifie as tert-butyldimethylsilyl derivatives (TBDMS-AA): AAs were dissolved in N-methyl-N-(t-butyldimethylsilyl) trifluoroacetamide (MTBSTFA) mixed with 1 % trimethylchlorosilane (TMCS) (Sigma-Aldrich Co. Ltd.) and acetonitrile and heated at 120 °C for 1 h. One µL of TBDMS-AA solution was injected into the GC through a GCCIII combustion interface (Thermo Fisher Scientific) TBDMS-AAs were separated on a DB5 column (30 m, 0.25 mm i.d., 0.25 µm fil thickness) with helium as a carrier gas. AA identificatio and quantificatio were performed using commercial mixtures of 20 proteinogenic AAs (Sigma Aldrich). Norvaline (Sigma-Aldrich Co. Ltd.) was added to plant samples before hydrolysis as an internal standard for quantification Due to the addition of non-labeled carbon by TBDMS, AA ^{13}C enrichment was subsequently calibrated for each AA (Shinebarger et al., 200). Briefl , this calibration was based on the in-

dependent measurement of ^{13}C of the TBDMS derivatives of the commercial AA and an additional set of four ^{13}C-labeled AA (Rubino et al., 2014). The calibration equation involves the number of carbon atoms added as TBDM and the isotopic composition of the latter. This isotopic composition term disappears in the calculation of the isotope excess (cf. Eq. 1 below). When multiple peaks were encountered for a single AA, the main or the best individualized peak was chosen for both quantificatio and isotope ratio determination. The isotope ratios were calculated using ammonium sulfate IAEA-N1 ($\delta^{15}N = 0.43 \pm 0.07$ ‰), IAEA-N2 ($\delta^{15}N = 20.41 \pm 0.07$ ‰) and polyethylene IAEA CH7 ($\delta^{13}C = -32.15 \pm 0.05$ ‰) as secondary standards. The sucrose standard IAEA CH6 used as a control yielded a mean value of 10.43 ‰.

Phytoliths were extracted from plants using a high-purity protocol based on acid digestion and alkali immersion steps previously described in detail by Corbineau et al. (2013). Phytolith samples were observed in natural light microscopy to determine their morphological assemblage and check for the absence of residual organic matter particles. An additional purity check was done via scanning electron microscopy (SEM) (Corbineau et al., 2013; Reyerson et al., 2015). Then, phytolith samples were analyzed for their C and N contents, as well as their ^{13}C and ^{15}N abundances by EA (Fisons NA 1500NC) coupled to a continuous-fl w IRMS (Finnigan Delta Plus). About 6–10 mg of phytolith concentrates were weighed using a pre-calibrated microbalance (Sartorius AG, Göttingen, Germany) into tin capsules (5×9 mm capsules, Costech Analytical Technologies Inc., Valencia, CA, USA) and pre-baked at 100 °C for 2 h to remove extraneous contaminants. To assure accurate integration and linearization of the raw analytical data obtained from the lower C and N peaks, we decreased the helium carrier fl w rate and measured several aliquots of in-house collagen L-cystine 99 % ($\delta^{13}C = -28.74 \pm 0.13$ and $\delta^{15}N = -6.14 \pm 0.07$ ‰; from Sigma Aldrich Co. Ltd) and atropine ($\delta^{13}C = -21.30 \pm 0.06$ and $\delta^{15}N = -2.90 \pm 0.10$ ‰; from Costech 031042) as well as the internationally certifie reference materials (e.g., USGS24 graphite $\delta^{13}C = -16.05 \pm 0.07$ ‰ and ammonium sulfate – IAEA-N1 $\delta^{15}N = +0.43 \pm 0.07$ ‰). Aliquots of baked-clean silicon dioxide (SiO_2; mesh no. −325, Sigma Aldrich, St. Louis, MO, USA) and fossil phytoliths (MSG70; Crespin et al., 2008) were also analyzed to provide independent blank data (Santos et al., 2010) and to check accuracy. To serve as quality assurance, note that the reproducibility obtained on the phytolith laboratory standard MSG70 was SD ±0.2 ‰ for $\delta^{13}C$ and $\delta^{15}N$, and SD ±0.01 % for C and N.

2.4 Calculations

For the control tank (no labeling), results are reported as δ values in per mil (‰) relative to the Vienna Pee Dee Belemnite (V-PDB) for $\delta^{13}C$ and in permil (‰) relative of the atmospheric N_2 for $\delta^{15}N$. For the labeled tanks. AA-derived ^{13}C and AA-derived ^{15}N plant concentration, recovery and net uptake were calculated on the basis of ^{13}C excess and ^{15}N excess of a sample relative to the control tank samples according to Eq. (1):

$$^{13}\text{C excess}_{\text{sample}}(\%) = {}^{13}\text{C atom}_{\text{sample}}(\%) - {}^{13}\text{C atom}_{\text{control}}(\%), \quad (1)$$

where $^{13}C\ atom_{sample}$ is the ^{13}C atom abundance of a sample (stems/leaves, roots or phytoliths) from a labeled tank and $^{13}C\ atom_{control}$ is the ^{13}C atom abundance of the same sample from the control tank.

The concentration of AA-derived ^{13}C in a sample, expressed in percent (%) of total C in the sample, was calculated using Eq. (2):

$$\text{AA-derived}^{13}\text{C}_{\text{sample}}(\%) = {}^{13}\text{C excess}_{\text{sample}}(\%) / \left({}^{13}\text{C atom}_{\text{solution}}(\%) - {}^{13}\text{C atom}_{\text{control}}(\%)\right) \times 100. \quad (2)$$

Recovery of AA-derived ^{13}C in a sample, expressed in micrograms per gram ($\mu g\,g^{-1}$) of the dry matter weight (d wt), was calculated using Eq. (3):

$$\text{Recovery AA-derived}^{13}\text{C}\left(\mu g g^{-1}\right) = \text{AA-derived}^{13}\text{C}_{\text{sample}} \times [\text{C}]_{\text{sample}}\left(\mu g g^{-1}\right) \quad (3)$$

where $[C]_{sample}$ is the concentration of total C in the sample.

Net uptake of AA-derived ^{13}C, expressed in percent (%) of $AA^{13}C$ initially supplied to the solution, was calculated using Eq. (4):

$$\text{AA-derived}^{13}\text{C uptake} = (\%)\,\text{recovery AA-derived}\ ^{13}\text{C}\left(\text{mg g}^{-1}\right) \times \text{d wt}_{\text{sample}}(\text{g}) / \text{AA}^{13}\text{C}_{\text{max supplied}}(\text{mg}), \quad (4)$$

where $d\,wt_{sample}$ is the dry weight of sample and $AA^{13}C_{max\ supplied}$ is the maximum $AA^{13}C$ supplied to the solution (322 mg of AA ^{13}C).

The same calculations were applied to ^{15}N.

3 Results

3.1 Total C, N and AA concentrations in the plants

After 14 days in the growth solution, the aboveground part of the plants of *Festuca arundinacea* were 30cm high but had not reached maturity or fl wer development (Fig. 1). The C / N ratios of the stems and leaves were similar in the labeled and control plants (8.4 and 9.0), whereas the C / N ratios of roots were higher in labeled (11.0) than in control

(4.2) plants (Table 1; Fig. 2). Recovered AAs accounted for 52 mg C g^{-1} of the dry matter in both roots and leaves. This is a high AA content, also attested by the low C / N ratio, in agreement with the high N level requirement of young plants sufficientl fertilized to support rapid protein synthesis (Mattson, 1980). However AA-derived ^{13}C and AA-derived ^{15}N only accounted for ca. 13–14 % of total C and 40 % of total N. This N contribution was lower than what might be expected. Indeed, at any stage of growth, AAs (in the form of proteins or free molecules) should account for more than 50 % of grass N. This discrepancy can be attributed to an underestimation of AAs by the extraction–purificatio process (i.e., incomplete hydrolysis recovery due to recombination into strong acids, and incomplete silylation). An underestimation of the AAs is consistent with the fact that amino-acid TBDMS derivatives, such as tryptophane or cystine (the cystine dimer), could not be recovered. However such an underestimation should not bias the measured relative proportion of methionine, phenylalanine and alanine.

3.2 Excess, uptake and recovery of AA-derived ^{13}C and AA-derived ^{15}N in the plants

The δ values of the roots and aerial parts of the control plants were respectively −31.0 and −31.7 ‰ for $\delta^{13}C$ and 13.8 and 14.5 ‰ for $\delta^{15}N$ (Table 1). These values were in the range of the ones measured for C_3 grasses in natural conditions ($\delta^{13}C$ from −22 to −34 ‰; e.g., O'Leary, 1988), ensuring that CO_2 potentially produced by decomposition of the ^{13}C-labeled molecules inside the tanks did not contaminate the growing chamber atmosphere. The amount of labeled C recovered in plants and phytoliths was thus considered as exclusively resulting from root uptake.

The low number of replicates (two labeled tanks and two molecular extracts per sample) was a compromise basically constrained by the large amount of matter required to isolate phytolith-occluded C and the experimental/analysis/cost effort. Table 1 shows that standard deviations calculated on the two replicates were always 1 order of magnitude lower than the mean values.

Relative to plants from the control tank, plants from the labeled tanks were enriched by 0.05 in ^{13}C and 0.5 % in ^{15}N in the roots and by 0.02 in ^{13}C and 0.3 % in ^{15}N in the stems and leaves (Table 1).

Overall, the net uptake of AA-derived ^{13}C and AA-derived ^{15}N by the plant represented respectively 4.5 and 46.9 % of the $AA^{13}C$ and $AA^{15}N$ added to the solution (Table 1). Whereas 35 % of absorbed AA-derived ^{13}C and 19.2 % of absorbed AA-derived ^{15}N were stored in the roots, 64.4 and 81.1 % of the absorbed AA-derived ^{13}C and AA-derived ^{15}N, respectively, were allocated to the stems and leaves (after Table 1). The associated AA-derived (^{13}C / ^{15}N) ratios were 0.8 in the roots and 0.3 in the stems and leaves (Table 1, Fig. 2).

Concentrations of AA-derived ^{13}C and AA-derived ^{15}N represented only 0.13 of total C and 2.8 % of total N of the plant (Table 1; Fig. 2). This contribution was higher in roots (0.28 of C and 4.1 % of N) than in stems and leaves (0.10 of C and 2.6 % of N). In roots, AA-derived ^{13}C was more concentrated in AAs than in total plant matter (0.70 vs. 0.28 % of C) (Table 1). When translocated to leaves, AA-derived ^{13}C concentration in AAs decreased to reach that of the bulk leaf matter (0.10 % of C) (Table 1).

Among the measured AAs in plant, alanine and phenylalanine were more abundant by more than a factor 10 relative to methionine (Table 1). However, alanine was not more enriched in ^{13}C than most of the AAs (Fig. 3). Instead, phenylalanine and methionine were significantl more enriched in ^{13}C than other AAs in roots, stems and leaves (Fig. 4).

3.3 Concentrations of phytoliths, phytC, phytN, AA-derived ^{13}C and AA-derived ^{15}N in phytC and phytN

Silica content measured by ICP-AES accounted for 0.08 and 0.26 % of the dry weight of roots and stems/leaves, respectively, which is lower than the > 1 % d wt concentration previously measured for this species harvested 8 weeks after sowing (Hartley et al., 2015). This is possibly due to the fact that the plants did not reach maturity and/or the volume of the roots in contact with the Si-enriched solution was small, limited by the RHYZOtest configuration Most of the stems' and leaves' silica was in the form of phytoliths (0.19 % d wt) (Table 2), which constituted a morphological assemblage characteristic of the Festucoideae grass subfamily to which *Festuca arundinacea* belongs (Honaine et al., 2006) (Fig. 4). As expected, root phytoliths were not abundant enough to be quantified PhytC represented 0.51 % d wt of phytoliths (Table 2), which is in the range of values previously measured for phytC (Santos et al., 2010; Alexandre et al., 2015; Reyerson et al., 2015). Occluded N (phytN) accounted for 0.10 % d wt of phytoliths.

Phytoliths were slightly more enriched in ^{13}C (^{13}C excess of 0.026 ± 0.002 %) than the leaves (^{13}C excess of 0.017 ± 0.001 %) (p value < 0.05) (Table 1). The AA-derived ^{13}C concentration in phytoliths represented 0.15 ± 0.01 % of phytC, which is low but on the same order of magnitude as the concentrations in the bulk matter and AAs of stems and leaves (0.10 ± 0.003) (Table 1). The AA-derived (^{13}C / ^{15}N) ratio in phytoliths was low (0.8) but higher than in bulk stems and leaves (0.3) (Table 1; Fig. 2).

4 Discussion

4.1 Plausible forms of AA-derived ^{13}C and AA-derived ^{15}N absorbed and translocated

Festuca arundinacea may have absorbed AA-derived ^{13}C and AA-derived ^{15}N from the labeled solution in multiple organic and inorganic forms, as detailed below. The AA-derived (^{13}C / ^{15}N) ratios of roots and leaves that were much

Table 1. Allocation of AA-derived ^{13}C and AA-derived ^{15}N in *Festuca arundinacea* grown in labeled and control solutions. Mean values and standard deviations (numbers in brackets) are given. The numbers in bold refer to the percentage of the individual AA applied. Un. is for unanalyzed.

	Dry weight	Total elements			Isotopic composition				Label concentration		Recovery of label from solution			Label net uptake	
	d wt g	[C] mg g^{-1} d wt	[N] C / N	‰	$\delta^{13}C$ atom %	^{13}C excess ‰	$\delta^{15}N$ atom %	^{15}N excess % plant C	[AA $^{13}C_{sample}$] % plant N	[AA $^{15}N_{sample}$] µg C g^{-1} d wt	Recovery AA ^{13}C µg N g^{-1} d wt	Recovery AA ^{15}N	AA-^{13}C / ^{15}N % supplied C	AA ^{13}C uptake %supplied N	AA ^{15}N uptake
Control tank															
Aerial part	16.2	389.4	43.4	9.0	31.7		13.8								
Phytolith	0.03	0.0	0.0	5.2	28.8		3.2								
Roots	4.4	270.3	63.9	4.2	31.0		14.5								
Labeled tank – aerial parts															
Total	23.9	395.0	46.8	4.9		0.017		0.338	0.10	2.6	385.0	1201.7	0.3	2.86	37.94
	(0.5)	(15.0)	(4.0)			(0.001)		(0.047)	(0.003)	(0.4)	(1.7)	(66.4)		(0.05)	(1.23)
Phytolith	0.040	5.1	1.05	4.9		0.026		0.118	0.15	0.9	0.02	0.0002	0.8	$2\ 10^{-7}$	$1\ 10^{-8}$
	(0.001)	(0.9)	(0.07)	(1.2)		(0.002)		(0.038)	(0.01)	(0.3)	(0.003)	(0.00006)		$(4\ 10^{-8})$	$(3\ 10^{-9})$
Sum AAs		52.5	13.2	4.0		0.020			0.10	N.D.	50.7	N.D.		**0.38**	Un.
		(6.4)	(1.6)			(0.009)			(0.000)		(0.0)			**(0.000)**	
Phenylalanine (PHE)		5.4	0.7	7.7		0.038			0.20	N.D.	12.1	N.D.		**0.18**	Un.
		(0.5)	(0.1)			(0.010)			(0.000)		(3.2)			**(0.047)**	
Methionine (MET)		0.3	0.1	4.3		0.187			1.10	N.D.	3.2	N.D.		**0.15**	Un.
		(0.2)	(0.0)			(0.027)			(0.200)		(0.5)			**(0.022)**	
Alanine (ALA)		4.7	1.8	2.6		0.012			0.07	N.D.	3.3	N.D.		**0.07**	Un.
		(1.1)	(0.4)			(0.001)			(0.000)		(0.3)			**(0.006)**	
Labeled tank – roots															
Total	4.9	376.0	34.3	11.0		0.048		0.534	0.28	4.1	1065.0	1387.2	0.8	1.61	8.98
	(0.5)	(13.0)	(3.6)			(0.000)		(0.100)	(0.002)	0.8	(43.6)	(117.5)		(0.24)	(1.74)
Sum AAs		52.0	14.6	3.6		0.112			0.70	N.D.	347.0	N.D.		**0.53**	Un.
		(5.3)	(1.5)			(0.013)			(0.100)		(49.6)			**(0.075)**	
Phenylananine (PHE)		2.3	0.3	7.8		0.499			2.90	N.D.	66.9	N.D.		**0.20**	Un.
		(0.2)	(0.0)			(0.155)			(0.900)		(20.8)			**(0.063)**	
Methionine (MET)		0.3	0.1	4.3		0.546			3.20	N.D.	9.8	N.D.		**0.10**	Un.
		(0.1)	(0.0)			(0.049)			(0.300)		(0.9)			**(0.009)**	
Alanine (ALA)		3.8	1.5	2.6		0.101			0.60	N.D.	22.6	N.D.		**0.10**	Un.
		(0.2)	(0.1)			(0.005)			(0.000)		(1.2)			**(0.005)**	
Labeled tank – total															
Total plants	28.8	391.5	44.7	8.8		0.022		0.371	0.13	2.8	500.7	1232.3	0.4	4.5	46.9
		(14.1)	(4.1)				(0.000)	(0.060)	(0.001)	(0.4)	(22.0)	(79.0)		(0.197)	(3.0)

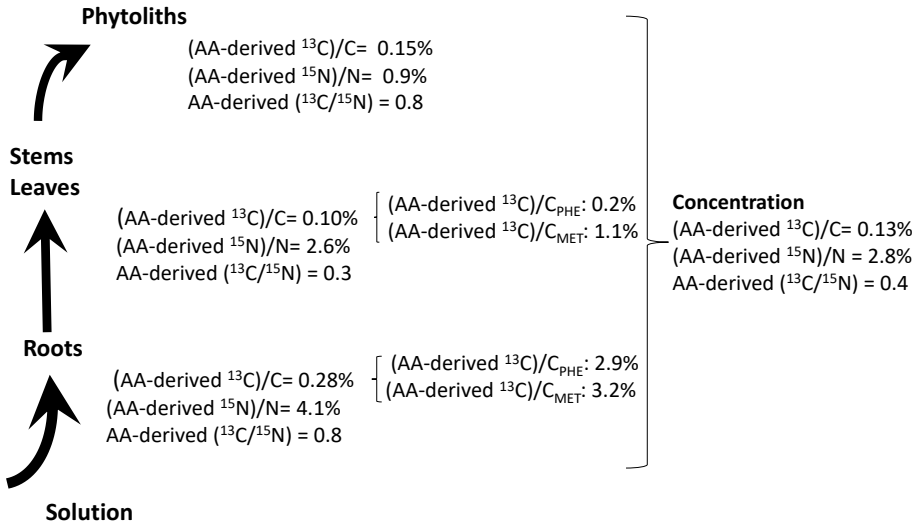

Figure 2. Concentration of AA-derived ^{13}C and AA-derived ^{15}N in bulk matter, phenylalanine (PHE) and methionine (MET) of roots, stems and leaves, and phytoliths of *Festuca arundinacea* grown in labeled tanks (in % of bulk C, N, PHE and MET).

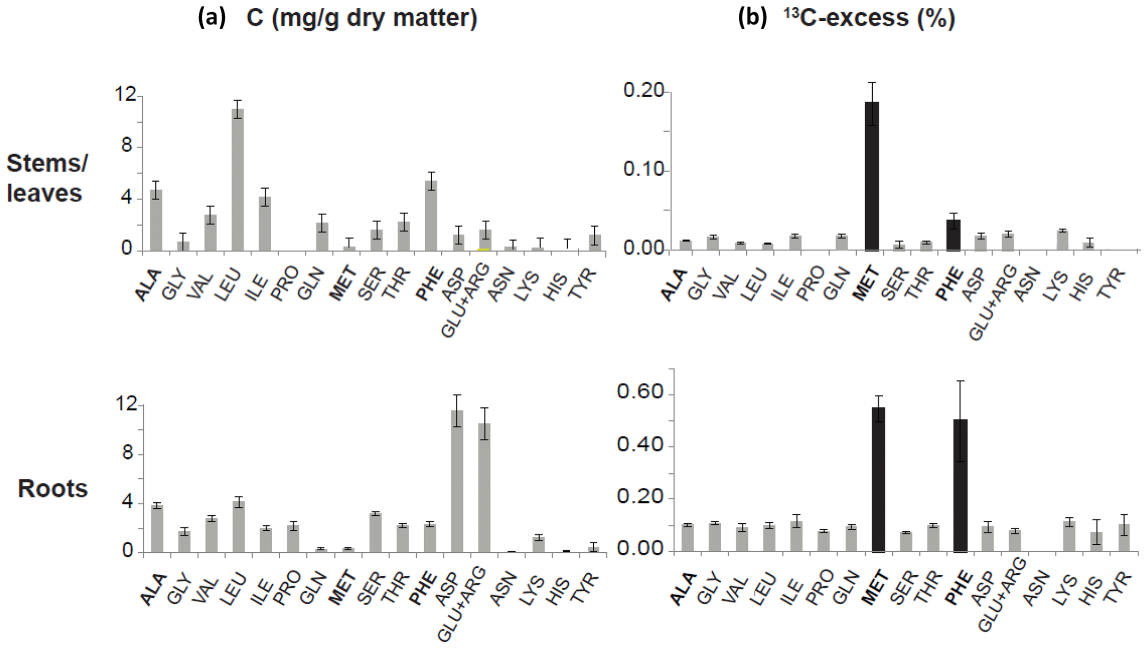

Figure 3. Concentration **(a)** and ^{13}C excess **(b)** of amino acids (AAs) measured by GC-IRMS in roots, stems and leaves. Bars stand for 1 standard deviation of four replicates (2 tanks × 2 AA extractions).

lower than C / N ratios of the supplied AAs (from 2.6 to 7.7) suggested that the grass absorbed most of ^{15}N from already mineralized N in the tank. Indeed, under non-sterile conditions, microbial activity around roots can biodegrade the AAs in a range of hours (Jones et al., 2005, 2009; Kielland et al., 2007) and produce derived metabolites plus mineralized N and CO_2 (Biernath et al., 2008; Rasmussen et al., 2010). Regarding C, both organic and inorganic forms may enter into the plant (Biernath et al., 2008; Rasmussen et al., 2010). Inorganic C can be transported through the plant passively, linked to transpiration (Vuorinen et al., 1989), and contribute to the C budget of the leaves through decarboxylation of the dissolved CO_2 and photosynthetic refixatio of released CO_2 (anaplerotic fixation Viktor and Cramer, 2005). Both organic and inorganic compounds can thus be used in the build-up of new molecules or as energetic resources (e.g., Näsholm et al., 2009), or be lost through respiration (Gioseff et al., 2012) or exudates (e.g., Jones and Darrah, 1993). Organic C can also

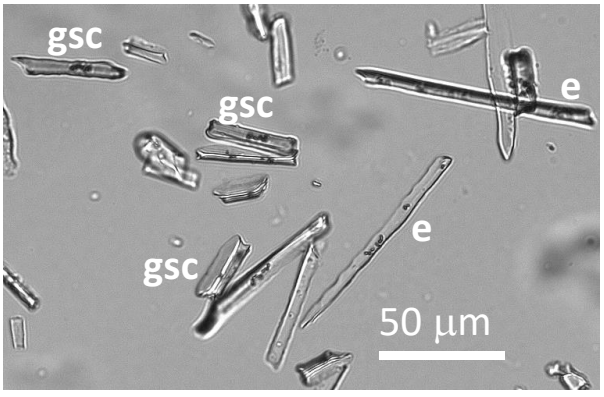

Figure 4. Natural light microscopy image of the phytolith assemblage produced by the stems and leaves of *Festuca arundinacea* dominated by (e) the elongate type and (gsc) the grass short cell trapeziform type (Madella et al., 2005).

Figure 4

Table 2. Concentration of phytoliths, phytolith occluded C (PhytC) and phytolith occluded N (PhytN) in *Festuca arundinacea* grown in labeled and control solutions. Numbers in italics refer to the standard deviation associated with the averaged values (one value per labeled tank).

	Phytolith	PhytC	PhytN
	% d wt plant	% d wt phytolith	
Control tank			
Aerial part	0.102	0.88	0.17
Roots	0.075		
Labeled tank			
Aerial parts	0.19	0.51	0.1
	(0.002)	(0.08)	(0.007)
Roots	0.014		
	(0.01)		

enter the plant as intact molecules, such as AAs (Sauheitl et al., 2009; Whiteside et al., 2009), and be either translocated by AA transporters or subject to deamination. Lastly, rhizospheric and endophytic microorganisms that acquired their labeled signature from the labeled solution may also account for the AA-derived ^{13}C and AA-derived ^{15}N recovery in the plant.

The present labeling experiment does not allow for precise tracing of the form in which the AA-derived ^{13}C and AA-derived ^{15}N were absorbed and fi ed in roots, stems and leaves, as recently done using a position-specifi C- and N-labeling technique (Moran-Zuloaga et al., 2015). All the processes described above may have occurred jointly. The significan decrease of ^{13}C excess (or AA-derived ^{13}C concentration) in AAs from roots to leaves (p value < 0.05) (Figs. 2 and 3) suggested either an uptake and fixatio of organic ^{13}C or an anaplerotic fixatio of inorganic ^{13}C in the roots themselves. However, the fact that ^{13}C was more concentrated in the extracted AAs than in bulk roots and stems/leaves (Table 1; Fig. 2) and, further, that ^{13}C excess values of methionine and phenylalanine were significantl higher than ^{13}C excess values of other AAs in the roots and stems/leaves (Fig. 3) evidenced that a small amount of AA^{13}C entered the plant and was subsequently translocated and fi ed in roots and stems/leaves in its original molecular form.

4.2 AA ^{13}C fixatio in phytoliths

In agreement with the radiocarbon evidence for soil C occlusion in phytoliths (Reyerson et al., 2015), AA-derived ^{13}C accounted for a measurable part of phytC. The phytolith C / N value (5.0) was close to a value previously measured in cultivated wheat phytoliths (3.7; Alexandre et al., 2015) and in the range of C / N values characteristic of AAs (4–5; Jones et al., 2009). However, the AA-derived ^{13}C / ^{15}N ratio (0.8) was far from this range. Thus, although our experiment tracked for the firs time the C from its absorption by grass roots to its fixatio in organic compounds subject to occlusion in stem and leaf phytoliths, the forms in which AA-derived ^{13}C entered the plant, was translocated and ultimately occluded in phytoliths still remain unknown.

Previous nano-scale secondary ion mass spectrometry (NanoSIMS) investigation of phytC indicated that at least part of phytC is continuously distributed in the silica structure, at the sub-micrometric scale (Alexandre et al., 2015). This has been further supported by Raman spectroscopy mapping (Gallagher et al., 2015). The process of silica precipitation has been investigated by environmental scanning electron microscope (ESEM) and transmission electron microscopy–energy-dispersive X-ray spectrometry (TEM-EDX) analyses, which have highlighted that silica firs precipitates in the inner cell wall, probably triggered by the presence of callose or lignin, and then infill the cell lumen in a centripetal way until most of the cell becomes silicifie (e.g., Perry et al., 1987; Motomura, 2004; Laue et al., 2007; Law and Exley, 2011; Zhang et al., 2013). During this process, an organic template probably participates in the silica formation (Harrison. 1996; Laue et al., 2007). When the cell silicificatio is complete, residual organic compounds that were not already occluded probably gather in any remaining spaces within the cell and delimitate micrometric central cavities characteristic of most phytoliths (Alexandre et al., 2015). In the present case, the concentration of AA-derived ^{13}C (relative to total C) in phytoliths, which is on the same order of magnitude as in leaves, supports a random fixatio of AA-derived ^{13}C in these residual organic compounds subject to occlusion in the silica structure. There are two plausible hypotheses for this fixation The firs hypothesis is that AA-derived ^{13}C may be associated with Si when absorbed by the roots, translocated in the plant and introduced into the cells. However, ^{29}Si NMR spectroscopy of ^{29}Si-labeled exudate of wheat xylem previously indicates only the occurrence of the dissolved forms of Si (Casey et al., 2004). Although

this does not preclude the subsequent formation of organo-silicate complexes, it weakens the hypothesis of Si and C being associated since their uptake by the roots. Additionally, in our experiment, the roots that contain the lowest amount of Si also contain the highest amount of AA-derived ^{13}C, which is not in agreement with AA-derived ^{13}C and Si being associated when absorbed by the roots. The second hypothesis is that AA-derived ^{13}C may be isolated as an unwanted substance in cell vacuoles and subsequently trapped in the silica structure. In order to check this hypothesis, we used D-ALA, expected to be less metabolized than L-AAs, although recent investigations suggest that plants are able to utilize D-AAs at rate comparable to those of other N forms (Hill et al., 2011). D-ALA was not specificall taken up or retained as an intact molecule (Fig. 3) and cannot account for the AA-derived ^{13}C measured in phytC. D-ALA may thus not be appropriate for tracing unwanted substances. Further investigations, including the use of spectroscopies relevant for characterizing phytC at the molecular level, are necessary to support or refute the above hypotheses.

4.3 Implication for our understanding of the C cycle at the plant–soil interface

In the experiments presented here, the net uptake of AA-derived ^{13}C by *Festuca arundinacea* represented 4.5 % of AA-derived ^{13}C supplied to the nutrient solution, part of it being absorbed as intact AA molecules (here methionine and phenylalanine). AA-derived ^{13}C fi ed in the plant represented only 0.13 % of total C, the root absorption of AA-derived ^{13}C being clearly marginal compared to photosynthesis. The present experiment was done in nutrient solution, and its relevance for soil C uptake assessment is therefore limited. It may underestimate the extent of the process under natural and fiel conditions. Indeed, AA uptake was shown to be inhibited by the high concentrations of mineral N preferred by the plant (Paungfoo-Lonhienne et al., 2008; Sauheitl et al., 2009; Gioseff et al., 2012). In the present case, mineral N may come from KNO_3 initially present in the original nutrient solution and from the supplied AAs dissociated by microbes, mycorrhizas or root exudates (Paungfoo-Lonhienne et al., 2008; Keiluweit et al., 2015). The maximum AA content in the growth solution (0.2 mmol L^{-1}) was higher than AA concentrations that have been measured in soil solutions (from 0 to 0.1 mmol L^{-1}; Vinolas et al., 2001; Jämtgård et al., 2010), thus potentially inhibiting AA uptake. Additionally, due the RHIZOtest configuration roots were confine to a small volume, and their contact with the renewed labeled solutions and AA-derived ^{13}C available for uptake was limited. Given the above considerations, the use of the AA-derived ^{13}C concentrations, experimentally measured, as a proxy of soil-derived C concentrations should be considered with caution.

However, to gain a rough idea on the order of magnitude of the C flu that may occur from soil to plant, at the ecosystem scale we used the 0.13 % AA-derived ^{13}C concentration in plants obtained in the present experiment and extrapolated that value to the grassland ecosystem scale. Grasslands cover a global surface of 2.4×10^9 ha (Scurlock and Hall, 1998) and are characterized by a net primary production (NPP) ranging from 7 to 20×10^9 t C ha^{-1} yr^{-1} (Scurlock et al., 2002). The global grassland productivity thus ranges from 16.8 to 48×10^9 t C yr^{-1}. The obtained flu of AA-derived C absorbed by grasses then would range from 21.8 to 62.4×10^6 t C yr^{-1}. This is nonsignifican when compared to the 2.6×10^9 t C yr^{-1} estimate for the land C sink (IPCC Staff, 2007), or to the 0.4×10^9 t C yr^{-1} estimate for the global long-term soil C accumulation rate (Schlesinger, 1990). It is, however, higher than a possible CO_2 phytolith biosequestration flu (e.g., Parr and Sullivan, 2005; Parr et al., 2010; Song et al., 2014). The CO_2 phytolith biosequestration concept is based on the assumptions that phytC is exclusively derived from atmospheric CO_2 and has a long residence time in soils. A recent re-examination of the CO_2 biosequestration flu by phytoliths, in the light of a lower and more realistic estimates of phytolith residence time in soils, yielded a value of 4.1×10^4 t C yr^{-1} for grasslands worldwide (Reyerson et al., 2015). The present study further minimizes the significanc of CO_2 biosequestration by phytoliths, showing that it could be counteracted by the flu of C potentially mobilized from soils by grass root uptake.

Recent experiments have demonstrated that root exudates promote a net loss of soil C previously assumed to be stable at the millennial scale thanks to its protection by mineral constituents (e.g., clays or amorphous minerals). Root exudates would stimulate microbial and fungi digestion (priming effect) (Fontaine et al., 2003, 2011) and promote dissolution of the mineral phase through an oxalic acid production experiment; we suggest that direct uptake of soil-derived C by roots, in conjunction with the N uptake, should be accounted for when investigating the role of roots in soil C mobilization.

5 Conclusions

In agreement with previous studies, the present labeling experiment supports the findin that C absorbed by grass roots and allocated to stems and leaves preserves to a small extent its original organic molecular form (here methionine and phenylalanine). Moreover, the experiment shows for the firs time that AA-derived C absorbed by grass roots and allocated to stems and leaves can partly feed the C ultimately fi ed in organic compounds subject to occlusion in stem and leaf silica. Further analyses are required to identify the form in which AA-derived C and more generally phytC is occluded. Our finding complement previous radiocarbon evidence of soil C contribution to phytC (Santos et al., 2012; Reyerson et al., 2015) and raise questions about the mechanisms that drive soil C mobilization by plant roots, which need to be an-

swered for a better understanding of soil–plant interactions involved in the terrestrial C cycle.

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