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Fate of rice shoot and root residues, rhizodeposits, and microbe-assimilated carbon in paddy soil: I. Decomposition and priming effect

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Abstract. The input of recently photosynthesized C has significant implications on soil organic carbon sequestration, and in paddy soils, both plants and soil microbes contribute to the overall C input. In the present study, we investigated the fate and priming effect of organic C from different sources by conducting a 300-d incubation study with four different ^{13}C-labelled substrates: rice shoots (Shoot-C), rice roots (Root-C), rice rhizodeposits (Rhizo-C), and microbe-assimilated C (Micro-C). The efflux of both $^{13}CO_2$ and $^{13}CH_4$ indicated that the mineralization of C in Shoot-C-, Root-C-, Rhizo-C-, and Micro-C-treated soils rapidly increased at the beginning of the incubation and then decreased gradually afterwards. In addition, the highest level of C mineralization was observed in Root-C-treated soil (45.4%), followed by Shoot-C- (31.9%), Rhizo-C- (7.9%), and Micro-C-treated (7.7%) soils, which corresponded with mean residence times of 33.4, 46.1, 62.9, and 192 d, respectively. Furthermore, the cumulative mineralization of native soil organic carbon in Shoot-C-treated soils was 1.48- fold higher than in untreated soils, and the priming effect of Shoot-C on CO_2 and CH_4 emission was strongly positive over the entire incubation. However, Root-C failed to exhibit a significant priming effect, which suggests that it could potentially be used to mitigate CH_4 emission. Although the total C contents of Rhizo-C- (1.89%) and Micro-C-treated soils (1.9%) were higher than those of untreated soil (1.8%), no significant differences in total C emissions were observed. However, the ^{13}C emissions of Rhizo-C- and Micro-C-treated soils gradually increased over the entire incubation period, which indicated that soil organic C-derived emissions were lower in Rhizo-C- and Micro-C-treated soils than in untreated soil, and that rhizodeposits and microbe-assimilated C could be used to reduce the mineralization of native soil organic carbon and to effectively improve soil C sequestration. The contrasting behaviours of the different photosynthesized C substrates suggests that recycling rice roots in paddies is more

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38 beneficial than recycling shoots and reveals the importance of increasing rhizodeposits and
39 microbe-assimilated C in paddy soils *via* nutrient management.
40
41 **Keywords:** Paddy soil; Rice; Plant residues; Rhizodeposits; Microbe-assimilated carbon; CO_2 and
42 CH_4 emission; Priming effect
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1 Introduction

The soils of rice paddies, which cover an area of ~165 million ha worldwide, hold great potential for expanded C sequestration (Conrad et al., 2012; Ge et al., 2012; Lal, 2004), and the soil organic carbon (SOC) pools in agricultural systems, of which plant C is the primary substrate, are significantly affected by the input of crop residues (Weintraub et al., 2007). For example, after crops are harvested or die, aboveground biomass, such as straw, stubble, and other surface debris, contribute to annual C inputs (Lu et al., 2003), and photosynthesized C substrates are continuously released by rice plants as rhizodeposits, such as decaying roots, throughout the growing season (Lu et al., 2002, 2003). However, autotrophic soil microbes that assimilate CO_2 contribute to C sequestration in paddy soil, as well (Ge et al., 2013; Yuan et al., 2012a), and since C inputs promote microbial activity and native SOC decomposition (Ye et al., 2015) and the quantity and quality of such inputs influence microbe-mediated decomposition processes (Brant et al., 2006; Creamer et al., 2015), the quantification of different C substrates allocated to paddy soils and their respective effects on native SOC require further investigation.

The aboveground biomass and root systems of rice plants represent the only inputs of available organic C to paddy SOC (Johnson et al., 2006), the quantity and quality of which has been reported previously (Chen et al., 2014; Kisselle et al., 2001; Zhang et al., 2015). However, although aboveground biomass has been reported to make significant contributions to SOC sequestration (Lu et al., 2003), rice roots have been reported to contribute 1.5–3-fold more C to SOC than shoots (Hooker et al., 2005), and similarly, Molina et al. (2001) reported that the stalks and leaves of corn contribute 50% less C to SOC than the roots and rhizodeposits. The predominant contribution of crop roots to SOC can partly be explained by the chemical composition of roots, which includes cellulose and lignin, as well as by residue–soil interactions, such as aggregate formation, which physically protect organic C from biodegradation (Baumann et al., 2009; Johnson et al., 2006; Lu et al., 2003).

Previous studies have also reported that the rhizodeposits of rice account for ~17% of the photo-assimilates (Nguyen, 2003) that enter paddy soil, and that rice rhizodeposits include soluble exudates, root border cells, dead debris, and insoluble mucilage (Lu et al., 2003). In cereal crops, 10–25% of root exudates are incorporated into SOC, and rhizodeposits are thought to play a key role in C cycling and sequestration in plant–soil–microbe systems (Kuzyakov, 2002; Kuzyakov et al., 2003).

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In addition to the photosynthesized C substrates of plants, soil microbes are also able to assimilate CO_2 *via* the Calvin-Benson-Bassham cycle and, thus, can significantly contribute to the net uptake and assimilation of atmospheric CO_2, as well (Ge et al., 2013; Yuan et al., 2012b). In fact, the CO_2 uptake by phototrophic soil microbes has been reported to account for up to 0.36% of the total C fixed in rice paddy soils and 0.19% of the total C fixed in upland soils (Ge et al., 2013; Yuan et al., 2012b).

However, the effect of C input from different C sources on the balance and stability of SOC has received limited attention. For example, low-molecular-weight C substrates are protected from mineralization *via* sorption onto soil particles (Jones and Edwards, 1998), which contributes to the stability and sequestration of SOC, whereas the input of fresh organic C, such as green manure, straw, and rhizodeposits, promotes the decomposition of native SOC and results in the emission of CO_2 and CH_4 (Huo et al., 2013; Yuan et al., 2014c). In addition, different C substrates can also have stimulating or restraining effects on the mineralization of native SOC, which are known as positive or negative priming effects (PEs), respectively (Kuzyakov, 2010). Priming is often caused by the addition of substrates with relatively high C availability and nutrient contents, which results in increased microbial activity (Blagodatsky et al., 2010); however, such easily degraded compounds greatly enhance the decomposition of native SOC (Blagodatsky et al., 2007; Qiao, et al., 2014), compared with the effects of ryegrass, cellulose, or wheat straw, which have complex structures that are less available to microbes (Kuzyakov and Bol, 2006; Kuzyakov et al., 2000).

Accordingly, the quantity and quality of different C inputs, as well as their fate and PE in paddy soils, are globally important (Bastida et al., 2013; Johnson et al., 2006; Wang et al., 2015). Although numerous studies have estimated the fate of plant residues and rhizodeposits in paddy soils, to our knowledge, there is no comparative information on (1) the decomposition of different organic C sources, such as rice shoots and roots, rhizodeposits, and microbe-assimilated C; or (2) the effects different organic C sources on the mineralization of native SOC. We hypothesized that the decomposition of such material would decrease with the increasing complexity of substrate composition and that the PE of plant residues was stronger than that of rhizodeposits and microbe-assimilated C, owing to their relatively higher quantity and stability in the soil. To investigate these hypotheses by quantifying the contribution of different organic C sources to CO_2 and CH_4

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emission and by analysing their PE, we conducted a 300-d incubation study using ^{13}C-labelled rice plant residues, rhizodeposits, and microbe-assimilated C in paddy soils.

2 Materials and methods

2.1 Study site and soil sampling

The experimental rice field was located at the Changsha Research Station for Agricultural and Environmental Monitoring, Hunan, China (113°19′52″ E, 28°33′04″ N; 80 m above sea level), where the soil was a typical stagnic anthrosol, developed from highly weathered granite, and the climate was subtropical, with a mean annual temperature and rainfall of 17.5 ℃ and 1300 mm, respectively.

Moist soil samples were collected from the plough layer (0–20 cm) and sieved (<4 mm) to remove visible plant residues. The soil contained 18.1 g kg^{-1} organic C, 1.8 g kg^{-1} total N, and 0.4 g kg^{-1} total K and had a pH of 5.6 and a soil: water ratio (w/v) of 1: 2.5.

2.2 Production of ^{13}C-labelled substrates

Rice cultivation and $^{13}CO_2$ labelling were performed as described by Ge et al. (2012; 2013), with some modifications. Briefly, 60 pots were filled with 1 kg dry soil, and of these, 40 pots were each planted with three 30-d-old rice seedlings (*Oryza sativa* L. 'Zhongzao 39'), whereas the remaining 20 pots were unplanted. Weeds were removed manually.

For ^{13}C labelling, 30 pots (20 planted, 10 unplanted) were transferred to an automatically controlled gas-tight growth chamber (110 cm length, 250 cm width, 180 cm height) and exposed to $^{13}CO_2$-fumigation for 18 d (May 14–31, 2013), during the vegetative growth period (including the entire tillering stage). The growth chambers were placed in a rice field to ensure that the environmental conditions of the labelled and control plants would be identical for labelled plants and unlabelled controls , and the remaining 30 pots (20 planted, 10 unplanted), which served as controls for measuring natural ^{13}C abundance, were placed 10–15 m from the growth chambers. The surface of each planted pot was covered with black plastic sheeting, to prevent algal photosynthesis in the floodwater and to

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ensure that only the rice shoots were exposed to $^{13}CO_2$ (i.e., not phototrophic microbes in the soil or water), whereas the unplanted pots were left uncovered, so that the soils were directly exposed to $^{13}CO_2$ and so phototrophic soil microbes could assimilate atmospheric $^{13}CO_2$. All the pots were watered every few days, in order to maintain a water depth of 2–3 cm above the soil surface, until harvest.

The CO_2 concentrations of the growth chambers were measured using an infrared analyser (Shsen-QZD, Qingdao, China) and maintained at 360–380 μl L^{-1}. The $^{13}CO_2$ was generated by acidifying $Na_2{}^{13}CO_3$ (1.0 M, 99 atom % ^{13}C; Cambridge Isotope Laboratories, Tewksbury, MA, USA) with H_2SO_4 (0.5 M) in beakers that were placed inside the growth chambers. During the labelling period, $^{13}CO_2$ was only released when CO_2 concentrations fell below 360 μl L^{-1}, and at CO_2 concentrations >380 μl L^{-1}, the gas flow was diverted and passed through CO_2 traps (NaOH solution). An air-conditioning system was used to control the temperature inside the chamber within 1 ℃ of the ambient temperature in the rice field. Two fans continuously circulated the air in the growth chamber.

2.3 ^{13}C-labelled substrate collection

All the rice plants and soils were sampled destructively after 18 d of $^{13}CO_2$ labelling. Rice shoots were removed at their bases, whereas rice roots were separated from the soil by washing with deionized water, and both shoots and roots were dried at 60 ℃ for 48 h and then cut into <5 mm pieces. ^{13}C-labelled rhizodeposits were obtained by gently shaking moist soil from the roots of rice plants and were prepared for incubation by removing root debris and mixing thoroughly. Meanwhile, to obtain microbe-assimilated ^{13}C, we collected soil from ^{13}C-treated, unplanted pots and mixed it thoroughly.

2.4 Soil incubation

To determine the PEs of different C sources and the effect of different C substrates on CO_2 and CH_4 emission, we conducted a 300-d incubation study of paddy soils that had been supplemented with ^{13}C-labelled shoots, roots, rhizodeposits, or microbe-assimilated C. Five treatments were used: (1) unlabelled and unplanted paddy soil supplemented with ^{13}C-labelled shoot residue (Shoot-C), (2) unlabelled and unplanted paddy soil supplemented with ^{13}C-labelled root residue (Root-C), (3) ^{13}C-labelled soil containing ^{13}C-labelled rhizodeposits (Rhizo-C), (4) ^{13}C-labelled soil containing ^{13}C-labelled microbe-assimilated C (Micro-C), and (5) unlabelled and unplanted soil without

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supplementation (CK). Three additional treatments were used to determine the natural occurrence of ^{13}C: (1) unlabelled and unplanted paddy soil with unlabelled shoot residue, (2) unlabelled and unplanted paddy soil with unlabelled root residue, and (3) unlabelled and unplanted paddy soil with unlabelled rhizodeposits.

For the Shoot-C and Root-C treatments, 150 g (100 g dry weight equivalent) unlabelled, unplanted soil with a water content of 50% was homogenized with 0.6 g of labelled and dried shoot and root residue, respectively, with a final residue content of 6 g kg^{-1}. Subsequently, the samples were transferred to 500 ml serum bottles with 100 ml deionized water, to ensure a water layer of >1–2 cm, and the bottles were sealed with butyl rubber stoppers. For the Rhizo-C and Micro-C treatments, 150 g fresh soil containing either ^{13}C-labelled rhizodeposits (from rice roots) or ^{13}C-labelled microbe-assimilated C (from labelled, unplanted pots) were directly weighed into 500 ml serum bottles, respectively. Incubation was conducted at 25 ℃ in the dark for 300 d, with four replicates for each treatment. CH_4 and CO_2 concentrations of the headspace samples were collected at 1, 3, 5, 10 d and then every 10 d after sealing, the gas was collected using a gas-tight syringe and stored in pre-evacuated Exetainer glass bottles (Labco, High Wycombe, UK). After each sampling point, the serum bottle was ventilated for 10 min, and then sealed with butyl rubber stoppers.

2.5 Analytical methods

The C content of the soil and plant residues (shoots and roots) was determined using dry combustion with an elemental analyser (vario MAX; Elementar Analysensysteme GmbH, Germany), whereas the CH_4 and CO_2 concentrations of the headspace samples were measured using a gas chromatographer (Agilent 7890A, Agilent Technologies, USA) equipped with a thermal conductivity detector for measuring CO_2 and a flame ionization detector for measuring CH_4. In addition, the stable C isotope composition of soils and plant residues were analysed using an isotope ratio mass spectrometer coupled with an elemental analyser (FLASH 2000; Thermo Fisher Scientific, USA), whereas the stable C isotope composition of CO_2 and CH_4 in the headspace samples were analysed using the isotope ratio mass spectrometer coupled with a GasBench (Thermo Fisher Scientific).

2.6 Calculations and statistical analysis

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185 The $\delta^{13}C$ values of plant residues, rhizodeposits, microbe-assimilated C, soils, CO_2, and CH_4 were
186 calculated as follows:

187 $$\delta^{13}C\ (‰) = \frac{Rs - R_{PDB}}{R_{PDB}} \times 1000\ ‰, \quad (1)$$

188 where R_{PDB} is the $^{13}C/^{12}C$ ratio of the international Pee Dee Belemnite (PDB) standard (R_{PDB} =
189 0.0112372; Lu et al., 2003) and *Rs* is the sample $^{13}C/^{12}C$ ratio. In addition, *Atomic* ^{13}C *(%)* was
190 calculated as follows:

191 $$Atomic\ ^{13}C\ (\%) = \frac{(\delta^{13}C + 100)\, R_{PDB}}{(\delta^{13}C + 100)\, R_{PDB} + 1} \times 100\%, \quad (2)$$

192

193 and the incorporation of ^{13}C in plant residues, rhizodeposits, microbe-assimilated C, bulk soils, CO_2,
194 and CH_4 was calculated as follows:

195 $$^{13}C_{sample}\ (\%) = [(Atomic\ ^{13}C), L - (Atomic\ ^{13}C), UL] \times \frac{C_{sample}}{100}, \quad (3)$$

196 where *(Atomic* ^{13}C*), L* and *(Atomic* ^{13}C*), UL* are the percentages of *Atomic* ^{13}C in labelled and
197 unlabelled samples, respectively, and $^{13}C_{sample}$ and C_{sample} are the total ^{13}C and C content of each
198 sample.

199 The $^{13}CO_2$ and $^{13}CH_4$ efflux (%) were calculated as the increases in $^{13}CO_2$-C and $^{13}CH_4$-C,
200 respectively, as percentages of the ^{13}C input, within each sampling interval, whereas cumulative $^{13}CO_2$
201 and $^{13}CH_4$ emission (%) were calculated as the percentages of the ^{13}C input represented by the sum of
202 the ^{13}C in $^{13}CO_2$ and $^{13}CH_4$, respectively, at each sampling day, and the mineralization percentage of the
203 input ^{13}C was calculated as the sum of total ^{13}C in CO_2 and CH_4, at each sampling day, relative to the
204 initially added total ^{13}C.

205 The temporal dynamics of the cumulative mineralization ratios were described by fitting a first
206 order single exponential decay curve:

207 $$y = y_0 + a\,(1 - e^{-bx}), \quad (2)$$

208 where *y* is the percentage of ^{13}C emission from the labelled-substrate C; y_0 is the pool of
209 labelled-substrate C remaining in the soil; *a* is the initial amount of bioavailable labelled-substrate C

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pool; b is the mineralization rate of substrate C; and x is time (d). Mean residence time ($1/b$) and half-life ($\ln(2)/b$) were also calculated.

The end-member mixing model was used to calculate the fractions of SOC- (C_{SOC}) and plant residue-derived C (C_{shoot} and C_{root}), as described by Phillips and Gregg (2001) and Phillips et al. (2005). This model allows the combination of mass spectrometric and efflux measurements. The shoot-derived $^{13}CO_2$ emission ($^{13}CO_{2shoot\text{-}derived}$) was calculated as follows:

$$^{13}CO_{2shoot-derived} = \frac{\delta^{13}CO_{2shoot} - \delta^{13}CO_{2CK}}{\delta^{13}CO_{2shoot} - \delta^{13}CO_{2soil}} \times CO_{2shoot}, \quad (5)$$

where $\delta^{13}CO_{2shoot}$ and $\delta^{13}CO_{2CK}$ are the $\delta^{13}C$ values of CO_2 derived from C_{shoot} and C_{CK}, respectively; $\delta^{13}C_{shoot}$ and $\delta^{13}C_{soil}$ are the $\delta^{13}C$ values of C_{shoot} and C_{soil}, respectively; and CO_{2shoot} is the total CO_2 derived from C_{shoot}; and the shoot-derived $^{13}CH_4$ emission ($^{13}CH_{4shoot\text{-}derived}$) and the root-derived $^{13}CO_2$ and $^{13}CH_4$ emission ($^{13}CO_{2root\text{-}derived}$ and $^{13}CH_{4root\text{-}derived}$, respectively) were calculated similarly (Phillips and Gregg, 2001; Ye et al., 2015).

The PE of SOM on CO_2 and CH_4 emission was calculated as follows:

$$PE_t\ (\%) = \frac{Gas - Gas_{CK}}{Gas_{CK}} \times 100\%, \quad (6)$$

where PE_t is the PE at time t (d); Gas is the total amount of CO_2 and CH_4 derived from C_{shoot} and C_{root}; and Gas_{CK} is the total amount of CO_2 and CH_4 derived from C_{CK} (Ye et al., 2015).

Analysis of variance in conjunction with Duncan's multiple range test ($p < 0.05$) and correlation analysis were conducted using SPSS 17 (SPSS Inc., Chicago, IL, USA), and figures were created using Origin 8.5 (OriginLab, Northampton, MA, USA).

3 Results

3.1 CO_2 and CH_4 emission of carbon substrate-treated soils

The atomic ^{13}C recovered from the CO_2 emission of Shoot-C- and Root-C-treated soils increased sharply at the beginning of incubation, reached a peak (4.71 and 4.39%) after 10 d, and then slowly declined until the end of the incubation; and the atomic ^{13}C of CO_2 from Rhizo-C- and Micro-C-treated soils exhibited a similar pattern, but with lower percentages (Fig. 1a). In addition, both Shoot-C and

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Root-C had similar effects on the amount of atomic ^{13}C recovered from CH_4, however, the CH_4 was below the detection limit in Rhizo-C- and Micro-C-treated soils (Fig. 1b).

The $^{13}CO_2$ efflux rates also increased rapidly at the beginning of incubation, peaked after 20 d, and then decreased gradually (Fig. 1c); however, the efflux rates from Shoot-C- (0.71%) and Root-C-treated (0.66%) soils were higher than those of Rhizo-C- (0.11%) and Micro-C-treated (0.06%) soils. The $^{13}CH_4$ efflux rates exhibited similar patterns (Fig. 1d). Furthermore, the cumulative $^{13}CO_2$ and $^{13}CH_4$ emissions also increased linearly during the first 60 d of incubation, after which they increase slowly (Fig. 2). The total ^{13}C per 100 g soil was 11.4, 5.75, 1.61, and 0.49 mg in the Shoot-C, Root-C, Rhizo-C, and Micro-C treatments, respectively (Table 1), and the total $^{13}CO_2$ emissions accounted for 28.6 and 43.8% of the initial ^{13}C from Shoot-C and Root-C, respectively, and 7.9 and 7.7% of the initial ^{13}C in Rhizo-C and Micro-C (Fig. 2a). In contrast, the cumulative $^{13}CH_4$ emissions only accounted for 3.3 and 1.6% of the initial ^{13}C from Shoot-C and Root-C, respectively (Fig. 2b).

The cumulative mineralization of substrate-derived ^{13}C was more rapid at the beginning of the incubation and followed a single exponential model (Fig. S1), and at the end of the incubation, we found that the total mineralization percentage was highest in Root-C-treated soils (45.4%), followed by Shoot-C- (31.9%), Rhizo-C- (7.9%), and Micro-C-treated (7.7%) soils. In addition, the bioavailable ^{13}C in the Shoot-C- and Root-C-treated soils was 34.2 and 46.2%, respectively, which was 4–5-fold larger than that of the Micro-C- (9.7%) and Rhizo-C-treated (7.8%) soils, and the mean residence time (MRT) of the Shoot-C-, Root-C-, Rhizo-C-, and Micro-C-treated soils was 33.4, 46.1, 62.9, and 192 d, respectively (Table 2).

3.2 Priming effect of Shoot-C and Root-C on CO_2 and CH_4 emission

Over the entire incubation period, the cumulative emissions of CO_2 and CH_4 from the untreated soil was 1692 mg kg^{-1}, and the SOC-derived C emissions from the Shoot-C- and Root-C-treated soils were 2519 mg kg^{-1} and 1737 mg kg^{-1}, respectively, which was 1.49- and 1.03-fold that of the untreated soil (Fig. 3). In addition, the end-member mixing model used to partition SOC-derived CO_2-C and CH_4-C suggested that the mineralization of native SOC was promoted by the Shoot-C and Root-C treatments.

Furthermore, the PE of the Shoot-C treatment peaked at 351% after 20 d of incubation and decreased to 46% by the end of the incubation, whereas the PE of the Root-C treatment peaked at 39%

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after 50 d of incubation and then decreased to 0.8%. Thus, the positive PE of Shoot-C was clearly stronger than that of Root-C, especially since the PE of Root-C was insignificant (i.e., $p > 0.05$; Fig. 4).

3.3 Mineralization of soil organic carbon in Rhizo-C- and Micro-C-treated soils

The total C emissions of Rhizo-C- and Micro-C-treated soils increased significantly from 116 mg kg^{-1} and 81 mg kg^{-1} after 10 d of incubation, respectively, to 1754 mg kg^{-1} and 1785 mg kg^{-1} by the end of the incubation. The total C emission of Rhizo-C-treated soil was significantly higher than that of the Micro-C- and un-treated soil, during the first 200 d of incubation; however, no significant differences were identified at the end of the incubation (Fig. 5a). In addition, the total ^{13}C emissions derived from the Rhizo-C and Micro-C treated soils gradually increased over the entire incubation, and the total mineralization of ^{13}C in Rhizo-C-treated soils was significantly higher than that in Micro-C-treated soils ($p < 0.05$; Fig. 5b). However, the C-mineralization of neither the Rhizo-C- nor Micro-C-treated soils were significantly different than that of untreated soil, which suggested that the rhizodeposits and microbe-assimilated C had effect on the mineralization of native SOC.

4 Discussion

4.1 Mineralization of carbon substrates in paddy soil

The atomic ^{13}C and effluxes of both CO_2 and CH_4 from soils treated with ^{13}C-labelled substrates exhibited a rapid increase at the beginning of the incubation, followed by a slow decrease (Fig. 1), which indicated that microbes prefer fresh C substrates over native SOC (Yuan et al., 2012c), as has been reported by previous studies on the decomposition of fresh C substrates in both paddy and upland soils (Lu et al., 2003; Parshotam et al., 2000). In these systems, the initial rapid decomposition is due to the addition of easily degradable organic C, in the form of highly bioavailable compounds with low molecular weight. Then, after the exhaustion of labile C, more recalcitrant components, such as cutin and lignin, and mineral-stabilized SOC are utilized (Baumann et al., 2009). However, the transition could also involve an alteration in species dominance, with rapidly proliferating bacteria using more available compounds during the early stages of decomposition and slower-growing fungi using the more recalcitrant components during later stages (Baumann et al., 2009; Brant et al., 2006).

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293 Both CO_2 and CH_4 efflux are important components of the C cycle in paddy soils and represent a
294 major proportion of the C released by microbial decomposition (Yuan et al., 2012c), and the results of
295 the present study suggest that the mineralization of shoot- and root-derived ^{13}C was ~3-4 times higher
296 than that of rhizodeposit- and microbe-derived ^{13}C (i.e., Root-C > Shoot-C > Rhizo-C > Micro-C; Fig.
297 2). The present study also found that the percentage of Root-C-derived ^{13}C recovered from CO_2 was
298 1.6-fold higher than Shoot-C-derived ^{13}C, which indicated that root residue was more easily
299 decomposed, a conclusion that was also supported by the higher $^{13}CO_2$ efflux of Root-C-treated soils.
300 However, the C mineralization rates of Rhizo-C- and Micro-C-treated soils were much slower, and
301 the MRTs of Rhizo-C- and Micro-C-treated soils were 2–4-fold higher than those of Shoot-C- and
302 Root-C-treated soils, likely owing to the formation of mineral-associated organic matter during the
303 labelling period that was well protected from microbial degradation and had a slow turnover rate
304 (Basler et al., 2015; Mikutta et al., 2014; Saidy et al., 2012; Schurig et al., 2013). Furthermore, most of
305 the C in Rhizo-C- and Micro-C-treated soils was not mineralized to CO_2 but, instead, underwent
306 intensive internal recycling (Gunina and Kuzyakov, 2015; Knowles et al., 2010) and were stored as
307 living microbial biomass, and the resulting biomass was either stabilized as occluded particulate
308 organic matter and mineral-associated organic matter (Basler et al., 2015, Schurig et al., 2013) or was
309 incorporated into metabolic products, such as sugars, carboxylic acids, and amino acids, which were
310 incorporated into cell membranes, cell walls, or polymers (Apostel et al., 2015; Gunina et al., 2014).
311

312 **4.2 Effect of carbon substrates on native SOC mineralization**

313 In the present study, the emission of CO_2 and CH_4 by Shoot-C- and Root-C-treated soils during the first
314 50 d were mainly derived from plant residue C, after which the contribution of native SOC increased.
315 However, a positive PE was observed until the end of the incubation, with the exception of
316 Root-C-treated soils (Fig. 4). These results are supported by previous studies that have reported that the
317 initial phase of rapid decomposition was the result of adding easily degraded organic C and other
318 available nutrients that promote both microbial activity and SOC decomposition (Chen et al., 2014;).
319 The compounds decomposed during the slower phase were less available for microbial growth, and as a
320 result of C limitation, most of the available C was likely incorporated into cells and converted to
321 storage compounds, rather than used for growth or respiration (Lu et al., 2003; Brant et al., 2006).

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However, the extracellular enzymes generated to degrade recalcitrant C substrates might be more effective in decomposing SOC at later stages of incubation, leading to a positive PE (Chen et al., 2014). In addition, the two phases of exogenous C decomposition and the mechanisms of PE simultaneously influence the strength and extent of native SOC mineralization (Chen et al., 2014; Ye et al., 2015).

As sources of C that is stabilized by soil minerals, both Rhizo-C and Micro-C augmented the C content of paddy soil (1.89 and 1.9%, respectively) over that of untreated soil (1.8%) and also reduced native SOC decomposition, which suggests that they could be used to protect native SOC, increase the organic carbon storage of paddy soil, and even mitigate global warming (Ge et al., 2012; Li and Yagi, 2004, Shen et al., 2014). In the present study, we found that the C emissions of Rhizo-C- and Micro-C-treated soils were similar to those of untreated soil, which suggested that rhizodeposits and microbe-assimilated C input have no effect on native SOC mineralization at any time during the rice-growing season. However, the total Rhizo-C- and Micro-C-derived ^{13}C increased gradually over the incubation period, despite the small initial input during the 18-d labelling period, which implies that the soils' native SOC-derived C were smaller than those of untreated soil and that both Rhizo-C and Micro-C suppressed the mineralization of native SOC. This suppression probably occurred because rhizodeposits and microbe-assimilated C are predominantly composed of highly bioavailable compounds with low molecular weight (Lu et al., 2002) and, in the present study, likely underwent internal recycling *via* microbial metabolism during the incubation, as indicated by their relatively longer MRTs (Table 2; Gunina et al., 2014; Mikutta et al., 2014; Schurig et al., 2013).

5 Conclusions

In the present study, Root-C-treated soils exhibited the highest rate of C mineralization, followed by Shoot-C-, Rhizo-C-, and Micro-C-treated soils, whereas the opposite trend was observed for MRT, and by the end of 300-d incubation, both Shoot-C- and Root-C-treated soils exhibited higher total mineralization and positive PEs. Although plant residues are widely used for improving soil fertility, their contribution to SOC assimilation is inefficient, and their use also contributes to the emission of greenhouse gasses. However, the present study demonstrates that both rhizodeposits and microbe-assimilated C can reduce native SOC decomposition and may more effectively contribute to the stability and sequestration of soil C.

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Manuscript under review for journal Biogeosciences
Published: 9 May 2016

References

Apostel, C., Dippold, M., Kuzyakov, Y.: Biochemistry of hexose and pentose transformations in soil analyzed by position-specific labeling and ^{13}C-PLFA, Soil Biol. Biochem., 80, 199-208, 2015.

Basler, A., Dippold, M., Helfrich, M., Dyckmans, J.: Microbial carbon recycling: An underestimated process controlling soil carbon dynamics–Part 2: A C3-C4 vegetation change field labelling experiment, Biogeosciences, 12(21), 6291-6299, 2015.

Bastida, F., Torres, I. F., Hernández, T., Bombach, P., Richnow, H. H., García, C.: Can the labile carbon contribute to carbon immobilization in semiarid soils? Priming effects and microbial community dynamics, Soil Biol. Biochem., 57, 892-902, 2013.

Baumann, K., Marschner, P., Smernik, R. J., Baldock, J. A.: Residue chemistry and microbial community structure during decomposition of eucalypt, wheat and vetch residues, Soil Biol. Biochem., 41, 1966-1975, 2009.

Blagodatskaya, E., Blagodatsky, S. A., Anderson, T. H., Kuzyakov, Y.: Priming effects in Chernozem induced by glucose and N in relation to microbial growth strategies, Appl. Soil Ecol., 37, 95-105, 2007.

Blagodatskaya, E., Khomyakov, N., Myachina, O., Bogomolova, I., Blagodatsky, S., Kuzyakov, Y.: Microbial interactions affect substrates of priming induced by cellulose, Soil Biol. Biochem., 74, 39-49, 2014.

Blagodatsky, S., Blagodatskaya, E., Yuyukina, T., Kuzyakov, Y.: Model of apparent and real priming effects: Linking microbial activity with soil organic matter decomposition, Soil Biol. Biochem., 42(8), 1275-1283, 2010.

Brant, J. B. Sulzman, E. W. Myrold, D. D.: Microbial community utilization of added carbon substrates in response to long-term carbon input manipulation, Soil Biol. Biochem., 38 (8) 2219-2232, 2006.

Chen, R., Senbayram, M., Blagodatsky, S., Myachina, O., Dittert, K., Lin, X., Blagodatskaya, E., Kuzyakov, Y.: Soil C and N availability determine the priming effect: microbial N mining and stoichiometric decomposition theories, Global Change Biol., 20(7), 2356-2367, 2014.

386 Conrad, R., Klose, M., Yuan, Q., Lu, Y., Chidthaisong, A.: Stable carbon isotope fractionation,
387 carbon flux partitioning and priming effects in anoxic soils during methanogenic degradation of
388 straw and soil organic matter, Soil Biol. Biochem., 49, 193-199, 2012.
389 Creamer, C. A., de Menezes, A. B., Krull, E. S., Sanderman, J., Newton-Walters, R., Farrell, M.:
390 Microbial community structure mediates response of soil C decomposition to litter addition and
391 warming, Soil Biol. Biochem., 80, 175-188, 2015.
392 Ge, T. D., Wu, X. H., Chen, X. J., Yuan, H. Z., Zou, Z., Li, B. Z., Zhou, P., Liu, S. L., Tong, C. L.,
393 Brookes, P., Wu, J. S.: Microbial phototrophic fixation of atmospheric CO_2 in China subtropical
394 upland and paddy soils, Geochim. Cosmochim. Acta., 113, 70-78, 2013.
395 Ge, T. D., Yuan, H. Z., Zhu, H. H., Wu, X. H., Nie, S. A., Liu, C., Tong, C. L., Wu, J. S., Brookes,
396 P.: Biological carbon assimilation and dynamics in a flooded rice – Soil system, Soil Biol.
397 Biochem., 48, 39-46, 2012.
398 Glanville, H., Rousk, J., Golyshin, P., Jones, D. L.: Mineralization of low molecular weight
399 carbon substrates in soil solution under laboratory and field conditions, Soil Biol. Biochem., 48,
400 88-95, 2012.
401 Gunina, A., Dippold, M. A., Glaser, B., Kuzyakov, Y.: Fate of low molecular weight organic
402 substrates in an arable soil: From microbial uptake to utilisation and stabilisation, Soil Biol.
403 Biochem., 77, 304-313, 2014.
404 Gunina, A., Kuzyakov, Y.: Sugars in soil and sweets for microorganisms: Review of origin,
405 content, composition and fate, Soil Biol. Biochem., 90, 87-100, 2015.
406 Hooker, B., Morris, T., Peters, R., Cardon, Z.: Long-term effects of tillage and corn stalk return on
407 soil carbon dynamics, Soil Sci. Soc. Am. J., 69(1), 188-196, 2005.
408 Hu, L. L., Su, Y. R., He, X. Y., Wu, J. S., Zheng, H., Li, Y., Wang, A.: Response of soil organic
409 carbon mineralization in typical Karst soils following the addition of ^{14}C-labeled rice straw and
410 $CaCO_3$, J. Sci. Food Agric., 92(5), 1112-1118, 2012.
411 Huo, L. J., Ji, X. H., Wu, J. M., Peng, H., Zhu, J.: Effects of applications of exogenous organic
412 carbon on methane emissions and oxidizable organic carbon in paddy soil, Res. Agric. Modern.,
413 34(4), 496-501, 2013. (English abstract in Chinese)

Biogeosciences Discuss., doi:10.5194/bg-2016-86, 2016
Manuscript under review for journal Biogeosciences
Published: 9 May 2016

414 Jones, D. L., Edwards, A. C.: Influence of sorption on the biological utilization of two simple
415 carbon substrates, Soil Biol. Biochem., 30(14), 1895-1902, 1998.
416 Johnson, J. M. F., Allmaras, R. R., Reicosky, D. C.: Estimating substrate carbon from crop
417 residues, roots and rhizodeposits using the national grain-yield database, Agron. J., 98(3), 622-636,
418 2006
419 Kisselle, K. W., Garrett, C. J., Fu, S., Hendrix, P. F., Crossley Jr, D. A., Coleman, D. C., Potter, R.
420 L.: Budgets for root-derived C and litter-derived C: comparison between conventional tillage and
421 no tillage soils, Soil Biol. Biochem., 33(7-8), 1067-1075, 2001.
422 Knowles, T. D. J., Chadwick, D. R., Bol, R., Evershed, R. P.: Tracing the rate and extent of N and
423 C flow from ^{13}C, ^{15}N-glycine and glutamate into individual de novo synthesized soil amino acids,
424 Org. Geochem., 41(12), 1259-1268, 2010.
425 Kuzyakov, Y.: Priming effects: Interactions between living and dead organic matter, Soil Biol.
426 Biochem., 42(9), 1363-1371, 2010.
427 Kuzyakov, Y., Bol, R.: Substrates and mechanisms of priming effect induced in two grassland
428 soils amended with slurry and sugar, Soil Biol. Biochem., 38(4), 747-758, 2006.
429 Kuzyakov, Y., Friedel, J. K., Stahr, K.: Review of mechanisms and quantification of priming
430 effects, Soil Biol. Biochem., 32(11-12), 1485-1498, 2000.
431 Kuzyakov, Y., Leinweber, P., Sapronov, D., Eckhardt, K. U.: Qualitative assessment of
432 rhizodeposits in non-sterile soil by analytical pyrolysis, J. Plant Nutr. Soil Sci., 166(6), 719-723,
433 2003.
434 Kuzyakov, Y., Siniakina, S. V., Ruehlmann, J., Domanski, G., Stahr, K.: Effect of nitrogen
435 fertilisation on below-ground carbon allocation in lettuce, J. Sci. Food Agric., 82(13), 1432-1441,
436 2002.
437 Lal, R.: Soil Carbon Sequestration Impacts on Global Climate Change and Food Security, Science,
438 304(5677), 1623-1627, 2004.
439 Li, Z., Yagi, K.: Rice root-derived carbon input and its effect on decomposition of old soil carbon
440 pool under elevated CO_2, Soil Biol. Biochem., 36(12), 1967-1973, 2004.

441 Lu, Y. H., Watanabe, A., Kimura, M.: Contribution of plant-derived carbon to soil microbial
442 biomass dynamics in a paddy rice microcosm, Biol. Fertil. Soil., 36(2), 136-142, 2002.
443 Lu, Y. H., Watanabe, A., Kimura, M.: Carbon dynamics of rhizodeposits, root- and shoot-residues
444 in a rice soil, Soil Biol. Biochem., 35(9), 1223-1230, 2003.
445 Mikutta, R., Lorenz, D., Guggenberger, G., Haumaier, L., Freund, A.: Properties and reactivity of
446 Fe-organic matter associations formed by coprecipitation versus adsorption: Clues from arsenate
447 batch adsorption, Geochim. Cosmochim. Acta., 144, 258-276, 2014.
448 Molina, J. A. E., Clapp, C. E., Linden, D. R., Allmaras, R. R., Layese, M. F., Dowdy, R. H.,
449 Cheng, H. H.: Modeling the incorporation of corn (Zea mays L.) carbon from roots and
450 rhizodeposition into soil organic matter, Soil Biol. Biochem., 33(1), 83-92, 2001.
451 Nguyen, C.: Rhizodeposition of organic C by plants: mechanisms and controls, Agron., 23(5-6),
452 375-396, 2003.
453 Parshotam, A., Saggar, S., Searle, P. L., Daly, B. K., Sparling, G. P., Parfitt, R. L.: Carbon
454 residence times obtained from labeled ryegrass decomposition in soils under contrasting
455 environmental conditions, Soil Biol. Biochem., 32(1), 75-83, 2000.
456 Paul, S., Veldkamp, E., Flessa, H.: Differential response of mineral-associated organic matter in
457 tropical soils formed in volcanic ashes and marine Tertiary sediment to treatment with HCl,
458 NaOCl, and $Na_4P_2O_7$, Soil Biol. Biochem., 40(7), 1846-1855, 2008.
459 Phillips, D., Newsome, S.D., Gregg, J.: Combining substrates in stable isotope mixing models:
460 alternative methods, Oecologia, 144(4), 520-527, 2005.
461 Phillips, D. L., Gregg, J. W.: Uncertainty in substrate partitioning using stable isotopes, Oecologia,
462 127(2), 171-179, 2001.
463 Qiao, N., Schaefer, D., Blagodatskaya, E., Zou, X., Xu, X., Kuzyakov, Y.: Labile carbon retention
464 compensates for CO2 released by priming in forest soils, Global Change Biol., 20, 1943-1954,
465 2014.
466 Saidy, A. R., Smernik, R. J., Baldock, J. A., Kaiser, K., Sanderman, J., Macdonald, L. M.: Effects
467 of clay mineralogy and hydrous iron oxides on labile organic carbon stabilisation, Geoderma, 173,
468 104-110, 2012.

469 Schurig, C., Smittenberg, R. H., Berger, J., Kraft, F., Woche, S. K., Goebel, M. O., Heipieper, H.
470 J., Miltner, A., Kaestner, M.: Microbial cell-envelope fragments and the formation of soil organic
471 matter: a case study from a glacier fore field, Biogeochemistry, 113(1), 595–612, 2013.
472 Shen J. L., Tang H., Liu J., Wang C., Li Y., Ge T. D. Davey L. J., Wu J. S.: Contrasting effects of
473 straw and straw-derived biochar amendments on greenhouse gas emissions within double rice
474 cropping systems, Agr. Ecosyst. Environ., 188, 264–274, 2014.
475 Tian, J., Dippold, M., Pausch, J., Blagodatskaya, E., Fan, M., Li, X., Kuzyakov, Y.: Microbial
476 response to rhizodeposition depending on water regimes in paddy soils, Soil Biol. Biochem., 65,
477 195-203, 2013.
478 Wang, W., Lai, D. Y. F., Wang, C., Pan, T., Zeng, C.: Effects of rice straw incorporation on active
479 soil organic carbon pools in a subtropical paddy field, Soil Till. Res., 152, 8-16, 2015.
480 Weintraub, M., Scott-Denton, L., Schmidt, S., Monson, R.: The effects of tree rhizodeposition on
481 soil exoenzyme activity, dissolved organic carbon, and nutrient availability in a subalpine forest
482 ecosystem, Oecologia, 154(2), 327-338, 2007.
483 Ye, R., Doane, T. A., Morris, J., Horwath, W. R.: The effect of rice straw on the priming of soil
484 organic matter and methane production in peat soils, Soil Biol. Biochem., 81, 98-107, 2015.
485 Yuan, H. Z., Ge, T. D., Chen, C., O'Donnell, A. G., Wu, J. S.: Significant role for microbial
486 autotrophy in the sequestration of soil carbon, Appl. Environ. Microbiol., 78(7), 2328-2336, 2012.
487 Yuan, H. Z., Ge, T. D., Wu, X., Liu, S., Tong, C. L., Qin, H., Wu, M., Wei, W. X., Wu, J. S.:
488 Long-term field fertilization alters the diversity of autotrophic bacteria based on the
489 ribulose-1,5-biphosphate carboxylase/oxygenase (RubisCO) large-subunit genes in paddy soil,
490 Appl. Microbiol. Biotechnol., 95(4), 1061-1071, 2012.
491 Yuan, Q., Pump, J., Conrad, R.: Straw application in paddy soil enhances methane production also
492 from other carbon substrates, Biogeosciences, 11(2), 237-246, 2014.
493 Zhang, G., Yu, H., Fan, X., Liu, G., Ma, J., Xu, H.: Effect of rice straw application on stable
494 carbon isotopes, methanogenic pathway, and fraction of CH_4 oxidized in a continuously flooded
495 rice field in winter season, Soil Biol. Biochem., 84, 75-82, 2015.
496

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Tables

Table 1. The carbon (C) content, atomic ^{13}C, and total ^{13}C in the soil and photosynthesized C substrates after 18 d of ^{13}C-labelling.

	Bulk soil	Shoot-C	Root-C	Rhizo-C	Micro-C
C content (%)	1.80 ± 0.12	40.50 ± 2.13	28.60 ± 1.15	1.89 ± 0.12	1.90 ± 0.11
Atomic ^{13}C (%)	1.08 ± 0.02	5.78 ± 0.09	4.43 ± 0.07	1.16 ± 0.03	1.11 ± 0.02
Total ^{13}C (mg)	0	11.43 ± 0.52	5.75 ± 0.41	1.61 ± 0.06	0.49 ± 0.05

Bulk soil, unplanted and unlabelled soil; Shoot-C, paddy soil supplemented with ^{13}C-labelled shoot residue; Root-C, paddy soil supplemented with ^{13}C-labelled root residue; Rhizo-C, paddy soil supplemented with ^{13}C-labelled rhizodeposits; Micro-C, paddy soil supplemented with ^{13}C-labelled microbe-accumulated C.

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505 **Table 2.** The size of bioavailable labelled-substrate C pool, mean residence time (MRT), and half-life
506 of cumulative ^{13}C recovery in CO_2 and CH_4 in four different incubation treatments.

Treatment	Size (%)	MRT (d)	Half-life (d)	R^2
Shoot-C	34.2	33.4	23.2	0.99
Root-C	46.2	46.1	31.9	0.99
Rhizo-C	7.8	62.9	43.6	0.98
Micro-C	9.7	192	133	0.98

507 The size of bioavailable labelled-substrate C pool (% initial ^{13}C), MRT, and R^2 were calculated based
508 on Fig. 1S. Shoot-C, paddy soil supplemented with ^{13}C-labelled shoot residue; Root-C, paddy soil
509 supplemented with ^{13}C-labelled root residue; Rhizo-C, paddy soil supplemented with ^{13}C-labelled
510 rhizodeposits; Micro-C, paddy soil supplemented with ^{13}C-labelled microbe-accumulated C.

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511 **Figures captions**

512 **Figure 1.** Atomic ^{13}C (%) recovered from CO_2 (**a**) and CH_4 (**b**) emissions and the $^{13}CO_2$ (**c**) and $^{13}CH_4$
513 (**d**) efflux (% initial ^{13}C) over the 300-d incubation period. The inset in Fig. 1**c** shows the $^{13}CO_2$ efflux
514 (% initial ^{13}C) of Rhizo-C and Micro-C over the 300-d incubation period. Values and error bars
515 represent means ± SE (n = 4). Shoot-C, unlabelled paddy soil supplemented with ^{13}C-labelled shoot
516 residue; Root-C, unlabelled paddy soil supplemented with ^{13}C-labelled root residue; Rhizo-C, paddy
517 soil containing ^{13}C-labelled rhizodeposits; Micro-C, paddy soil containing ^{13}C-labelled
518 microbe-accumulated C.
519
520 **Figure 2.** Cumulative $^{13}CO_2$ (**a**) and $^{13}CH_4$ (**b**) emissions (% of initial ^{13}C) over the 300-d incubation
521 period. Values and error bars represent means ± SE (n = 4). Shoot-C, unlabelled paddy soil
522 supplemented with ^{13}C-labelled shoot residue; Root-C, unlabelled paddy soil supplemented with
523 ^{13}C-labelled root residue; Rhizo-C, paddy soil containing ^{13}C-labelled rhizodeposits; Micro-C, paddy
524 soil containing ^{13}C-labelled microbe-accumulated C.
525
526 **Figure 3.** Cumulative CO_2 and CH_4 emissions by Shoot-C- (**a**) and Root-C-treated (**b**) soils over the
527 300-d incubation period. The inset in Fig. 3**b** shows total C emission derived from SOC of
528 Root-C-treated soils and CK. Values and error bars represent means ± SE (n = 4). Shoot-C, ^{13}C-labelled
529 shoot residue; Root-C, ^{13}C-labelled root residue; SOC, soil organic carbon; CK, unlabelled and
530 unplanted soil without supplementation.
531
532 **Figure 4.** Priming effect (%) of ^{13}C-labelled plant residues over the 300-d incubation period. Values
533 and error bars represent means ± SE (n = 4). Shoot-C, unlabelled paddy soil supplemented with
534 ^{13}C-labelled shoot residue; Root-C, unlabelled paddy soil supplemented with ^{13}C-labelled root residue.
535
536 **Figure 5.** Total C (**a**) and ^{13}C (**b**) emission by ^{13}C-labelled rhizodeposit- and microbe-accumulated
537 C-treated soils over the 300-d incubation period. Values and error bars represent means ±SE (n = 4).
538 Different letters indicate significant differences at $p < 0.05$ (Duncan multiple range test). Rhizo-C,

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539 paddy soil containing ^{13}C-labelled rhizodeposits; Micro-C, paddy soil containing ^{13}C-labelled

540 microbe-accumulated C; CK, unlabelled and unplanted soil without supplementation.

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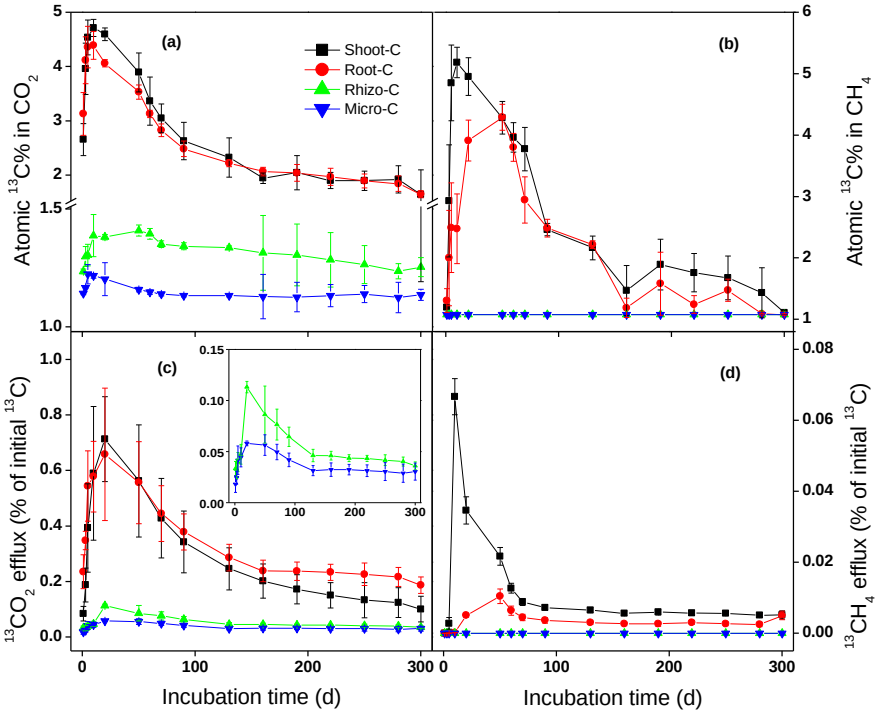
542 **Figure 1**



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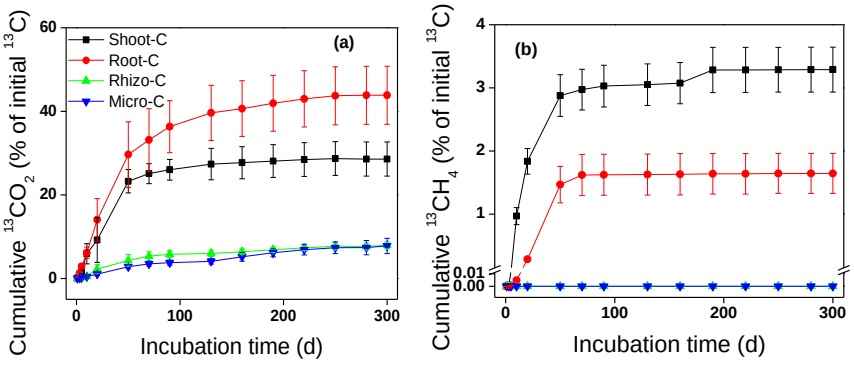
545 **Figure 2**



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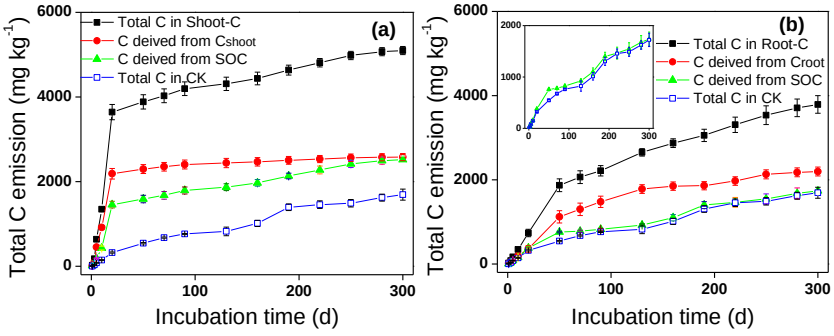
548 **Figure 3**



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Manuscript under review for journal Biogeosciences
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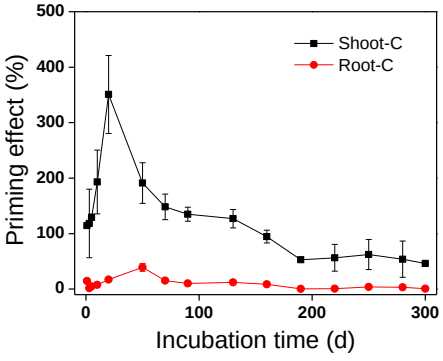
551 **Figure 4**



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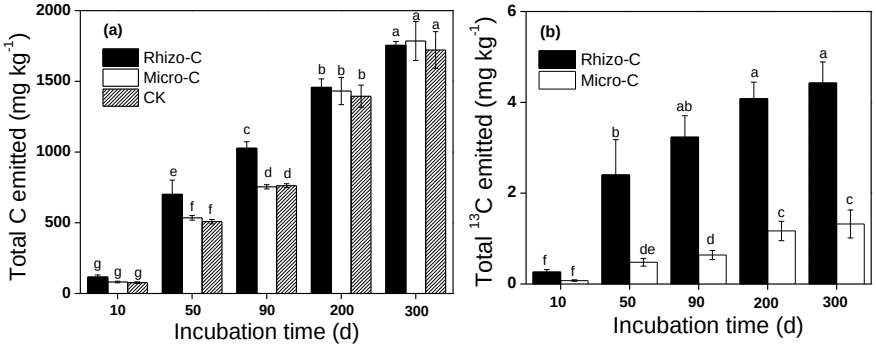
554 **Figure 5**



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