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Cesium transfer to millet and mustard as a function of Cs availability in soils

Flouret A., Henner P., Coppin F., Pierrisnard S., Carasco L., *Février L.

Authors affiliation: Institut de Radioprotection et de Sûreté Nucléaire (IRSN), PSE-ENV, SRTE/LR2T, B.P.3,
13115 Saint Paul-lez-Durance Cedex, France

* Corresponding author: laureline.fevrier@irsn.fr

Highlights

Soil/plant Cs transfer experiments on Rhizotests with three soils and two plants

The Cs CR of mustard is not always the highest, contrary to plant phylogeny predictions

Differences in CRs may partly relate to the translocation capacities of millet

Calculating CR based on the available Cs pool in soils reduce the range of CR variation

Keywords

Concentration Ratio, Soil/plant transfer, phylogeny, bioavailability, Rhizotest

Abstract

¹³⁷Cs is one of the most persistent radioactive contaminants in soil after a nuclear accident. It can be taken up by plants and enter the human food chain generating a potential human health hazard. Although a large amount of literature has highlighted the role of the different processes involved in Cs uptake by plants, there is still no simple way to predict its transfer for a specific plant from a particular soil. Based on the assumption that the concentration ratio (CR) of Cs can be predicted from one plant taxon if the CR of another taxon is known and taken as reference, whatever the supporting soils, a series of plant/soil Cs transfer experiments were performed on Rhizotest during 21 days using three soils with different textures, clay and organic matter contents and two plants (millet and mustard) with potentially contrasting Cs uptake capacity based on their phylogeny. CRs of each plant varied by 2 to 3 orders of magnitude depending on the soil and contrary to expectations, the CRs of mustard were either higher (for clay soil), equal (for clay-loam soil) or lower (for sandy soil) than the one of millet. Considering Cs availability in soils and defining a new CR based on the amount of Cs available in the soil (CR_{avail}) decreased the range of

29 variation in CR between the different soil types for a given plant by one order of magnitude. Differences
30 in Cs (and K) translocation to shoots, possibly specific to millet within Poales, could partly explain the
31 relative CRs of millet and mustard as a function of soils.

32

33 **1 Introduction**

34 After a nuclear accident like Chernobyl or Fukushima, soil is one of the compartments that receives a high
35 fraction of radioactive material during the fallout. From all the radionuclides discharged into the
36 environment, radiocesium (^{137}Cs) is one of most released radio-isotopes (Völkle et al., 1989; Hu et al.,
37 2010; Chino et al., 2011), which, in addition to its relatively long half-life, results in a long-term issue
38 regarding food chain transfers. After fallout and due to its high ability to accumulate in soils and sediments
39 (IAEA, 2010), ^{137}Cs is mostly found in the soil surface layer which represents the soil-root interaction zone
40 (Fujii et al., 2014; Jagercikova et al., 2015; Burger and Lichtscheidl, 2018; Takahashi et al., 2018). Thus, soil
41 is considered as a key compartment for the transfer of ^{137}Cs in the trophic chain (IAEA, 2010; Fesenko et
42 al., 2013). There is no known role for Cs in plants, but because of this similarity with potassium (K), Cs can
43 be absorbed from the soil pore water by the roots through the same pathway as K (Middleton et al., 1960;
44 White and Broadley, 2000; Zhu and Smolders, 2000; Qi et al., 2008). As ingestion of contaminated
45 agricultural products is one of the main components of human exposure (Rosén et al., 1995; Okuda et al.,
46 2013; Guillén et al., 2017), it is essential to predict the fate of ^{137}Cs throughout the soil-plant continuum.

47 In contaminated soil, transfer of Cs to plants depends on both the capacity of the soil to provide Cs to the
48 soil solution from where plants can take it up and the capacity of the plants to absorb it. Cs has been
49 shown to be strongly adsorbed at the surface of soil minerals or organic constituents (Absalom et al., 1995;
50 McKinley et al., 2001; Kruglov et al., 2008). Clays are generally considered as the main sorbent of Cs in soils
51 (Shenber, 1993; Qin et al., 2012; Hirose et al., 2015), through the involvement of several sorption sites,
52 such as "Frayed Edge Sites" (FES) or other cationic exchange sites (Sawhney, 1972; Brouwer et al., 1983;
53 Poinssot et al., 1999; Bradbury and Baeyens, 2000; Zachara et al., 2002; Missana et al., 2014a; Cherif et
54 al., 2017; Okumura et al., 2018). The role of organic matter in the adsorption of Cs in soils is still debatable.
55 Whereas it can play the role of sorbent in soils with high (>80%) organic matter content (Valcke and
56 Cremers, 1994; Rigol et al., 2002; Lofts et al. 2002), Valcke and Cremers (1994) showed that for soils
57 containing less than 40% of organic matter, the FES are the main sorption sites for Cs. Furthermore, a small
58 quantity of fulvic acid (2%) could decrease the sorption of Cs in soils due to a coating of fulvic acid on the
59 clay surface, impeding the sorption of Cs at the FES (Staunton and Roubaud, 1997). The same results were

60 observed with humic acid (Dumat and Staunton, 1999; Fan et al., 2014). Cs sorption on soil solid surfaces
61 can also be modified due to competitive interaction with other monovalent cations of the soil solution. In
62 particular, potassium (K) can reduce Cs adsorption by clays (Staunton and Roubaud, 1997; Missana et al.,
63 2014b), thus increasing its mobility and availability in soils.

64 When Cs is depleted from the soil solution (as can locally occur around plant roots taking Cs up), the re-
65 supply of the soil solution in Cs is driven by desorption processes from the soil solid phases. Desorption of
66 Cs from soil solid phases (clays or other) mainly depends on the surface on which Cs is adsorbed and on
67 the strength of the sorption. Consequently, Rigol et al. (1999) showed that for different soils the yield of
68 extractable ^{137}Cs with $1 \text{ mol.L}^{-1} \text{ CH}_3\text{COONH}_4$ varied from very few percent to 100% depending on soil nature
69 and number of extractions. In the same way, Teramage et al. (2018) observed that around 50% of ^{137}Cs in
70 a fresh contaminated soil was extractable with $1 \text{ mol.L}^{-1} \text{ CH}_3\text{COONH}_4$ whereas few tenths of percent was
71 extractable with ultra-pure water. Moreover, Valcke and Cremers (1994) observed a decrease in ^{137}Cs
72 extraction yield with desorption time. Recently, successive desorption in batch experiments has been
73 proposed as a method to estimate the pool of Cs irreversibly bound to the soil and the one that can be
74 transferred to the soil solution (Teramage et al., 2018; Coppin et al., in prep). This method offers the
75 opportunity to quantify the fraction of Cs in the soil that is available for re-supplying the soil solution, as
76 well as the strength of its binding within the soil independently of the mass/volume ratio used for the
77 extraction process.

78 Plant uptake of Cs by roots occurs by several pathways, not all of which have currently been identified.
79 However, as an analog to K, it has been shown that Cs may enter the plants through some of the K^+
80 transporter and K^+ channel pathways. Cs uptake is thus impacted by K homeostasis (ie. the ability of
81 plants to regulate their internal K concentrations at a steady and optimal level), and is regulated by factors
82 relating to external K concentration in the soil solution for root uptake strictly speaking and to K in the
83 plant (root and shoot stocks). Not all regulatory processes are already known, in particular *in planta*. Yet,
84 as for other nutrients, by sensing concentration at the solution/root interface and due to the complex
85 balance between plant demand for growth and the state of internal stocks, the nature and number of
86 active K transporters is permanently adapting, leading to constant variation in Cs transfer capacities.
87 Transfer through K^+ transporters is the preferred pathway at a low external concentration of K^+ (below
88 $100\text{-}300 \mu\text{mol L}^{-1}$ depending on the plant) whereas uptake through the K^+ channel dominates at a high
89 external concentration of K^+ . However, it should be noted that the latter is very specific to K^+ (and

90 discriminates significantly against cesium) while the former transport pathway shows little specificity to K
91 or Cs (Zhu and Smolders, 2000).

92 Therefore, increasing the level of K^+ in soil with initially low K contents can reduce Cs uptake by plants,
93 despite the fact that it will simultaneously favor Cs desorption from soil solid phases.

94 In radioecology, a simple and empirical approach that relates the concentration of Cs in the plant (shoot
95 or consumed organ, depending on the database) to the soil concentration through an aggregated
96 parameter, either called transfer factor - TF or concentration ratio – CR- depending on the authors, is often
97 used to predict plant uptake of Cs (Almahayni et al., 2019). Due to differences in plant ability to take Cs up
98 and differences in soil Cs sorption capacity, early efforts were made to propose TF values classified by
99 plant type and soil texture, with the underlying assumption that radionuclide availability particularly
100 depends on the nature of soil solid phases, imperfectly taken into account by soil texture classification.
101 With this approach, Nisbet and Woodman (2000) showed that TF values for brassicas and cereals were
102 higher for a sandy soil than for a clay soil. This classification is still the one proposed by the IAEA to predict
103 radionuclide transfer in terrestrial environments (IAEA, 2010). Other authors have tried to link the TF to
104 some soil properties resulting in models with a higher number of parameters. For example, TFs were
105 related to the concentration of exchangeable K in soils (Frissel et al., 2002; Kondo et al., 2014) or multiple
106 soil properties (clay content, amount of organic matter and amount of K in soils) (Absalom et al., 2001;
107 Tarsitano et al., 2011).

108 More recently, a new method based on plant phylogeny has been proposed, that could particularly help
109 to derive TF/CR for plants grown in all soil textures without having to perform experiments. This method
110 assumes that plant Cs absorption capacity is in part driven by plant phylogeny (Broadley et al., 1999; Willey
111 et al., 2005, 2010) and proposes a relationship between Cs plant concentration and plant phylogeny. As
112 an example, Eudicots were shown to exhibit significantly higher Cs concentrations (especially in the
113 Caryophyllales, Asterales and Brassicales) than Monocots (with Poales the lowest), independently of the
114 studied soils. The method implies that the relative differences in TF/CR for two plants belonging to two
115 significantly different taxa in terms of Cs uptake might, on average, be detectable across contrasting soil
116 types. Assuming that taxonomic position could be used to refine the prediction of Cs CRs, Beresford and
117 Willey (2019) and Beresford et al. (2020) developed the concept of a “benchmark-taxon” that can be used
118 to calculate CR values for plant taxa for which no data are available, based on the CR of a reference taxon
119 and the output of the Residual Maximum Likelihood (REML) analysis (REMLmean) provided in Beresford

120 and Willey (2019). The method relies on the assumption that on average the ratio between the CR of two
121 taxa is equal to the ratio of the REMLmean of these two taxons whatever the studied sites.

122 Therefore the goal of our study was to check if plants taken from two taxa chosen for their potential
123 differential ability for Cs transfer and grown in soils chosen to provide different Cs availability can give
124 valuable insights on how plant-soil interactions may influence the extent of Cs transfer. Experiments were
125 performed on a monocot and a dicot (from the Poaceae and Brassicaceae families respectively), with three
126 soils having different clay content, organic matter, and pH levels. Experiments were performed on a
127 Rhizotest, a tool specifically designed to assess the phytoavailability of contaminants in soils through root
128 uptake (Chaignon and Hinsinger, 2003; Bravin et al., 2010; Mihalík et al., 2012). In addition to the plant
129 uptake of Cs, the availability of Cs in soils and the concentration of K in the soil-solution and within the
130 plants were also assessed. The dynamics of all these parameters were monitored over 21 days of exposure.

131

132 **2 Material and methods**

133 2.1 Soils and plants studied

134 2.1.1 Soil properties

135 Three soils were selected for their contrasting physicochemical and mineralogical properties (Table 1). Soil
136 samples were air-dried and sieved to 2 mm before analysis. Soil physicochemical analyses were performed
137 by INRA LAS laboratory (Arras, France). Exchangeable cations and CEC were measured using the
138 ammonium acetate extractant method (NFX 31-130). N and organic matter content were measured using
139 a dry combustion protocol (ISO 13878: 1998 and ISO 10694: 1995 respectively) and CaCO₃ content was
140 measured by a volumetric method (ISO 10693: 1995). Soil mineralogy of the fraction below 2 µm was
141 determined by ERM laboratory (Poitiers, France) using a powder X-ray diffractometer (Bruker D8 Advance
142 A25) with CuKα radiation at 40 kV and 40 mA on oriented mounts. The CEC of the 2µm fraction was
143 measured by BRGM laboratory (Orléans, France) with the same technique as the CEC of the bulk soil.

144 2.1.2 Plants

145 Two plants were chosen based on their potential different Cs uptake capacities as determined in the
146 phylogenetic analysis of Willey et al. (2005): mustard (*Brassica juncea*), belonging to the Brassicaceae
147 family, and millet (*Panicum millaceum*), belonging to the Poaceae family according to the AGP III
148 taxonomic classification. Plants were purchased from “Les Semences du Puy”, Le Puy-en-Velay, France.

149

150 2.2 Soil-plant transfer experiment in Rhizotest

151 2.2.1 Experimental set up

152 Plants were exposed to Cs contaminated soils in a Rhizotest design (Mihalík et al., 2012; Henner et al.,
153 2018); inspired by the RHIZOtest developed as a normative experimental tool to measure trace element
154 bioavailability (ISO 16198:2015). This device (Figure S1) is composed of 2 parts: the upper one, closed at
155 its base by a 30- μm permeable nylon mesh membrane, on which the plant develops a root mat during a
156 hydroponics growing step, and the lower one, which contains a thin contaminated soil layer (≈ 5 mm thick).
157 Both parts are put into close contact during the exposure step. The mesh physically, but not chemically,
158 separates the soil and plant compartments. This mesh prevents root contamination by soil particles, and
159 thus facilitates the collection of clean roots at the end of the experiment.

160 Each experiment lasted 46 days and could be subdivided into two steps:

- 161 • the growing step (25 days): plants develop on the upper part of the device in hydroponic
162 conditions with a complete nutrient solution. At the end of this step, 100% of the membrane area
163 is covered by a dense root mat, which is a prerequisite for the test.
- 164 • the soil exposure step (maximum 21 days) in which the 2 parts are in contact.

165 Six experiments, each defined by a combination of one soil (E, H and S) and one plant (mustard or millet),
166 were conducted, one at a time in triplicate. Each experiment consisted of (Figure S1-b):

- 167 • planted and unplanted (control) Rhizotests;
- 168 • 6 sampling dates for planted Rhizotests (day 2, 3, 4, 7, 14 and 21) and 5 sampling dates for
169 unplanted Rhizotests (day 2, 4, 7, 14, 21).

170 On day 0, 3 upper and 3 lower parts were sampled for the initial characterization of the plant and soil
171 respectively. Therefore, in total, 21 upper parts with plants and 36 lower parts with soil were prepared for
172 each experiment.

173 The experiment was conducted in a growth chamber (Fitotron SGC 120, Weiss) with the following
174 program: 16 h/8 h light/night cycle, $25/20 \pm 1$ °C day/night temperature, $70 \pm 5\%$ relative air humidity and
175 light intensity of $200 \mu\text{mol m}^{-2} \text{s}^{-1}$. Rhizotests were randomly placed in the growth chamber and their
176 positions were randomly moved each day to avoid any bias due to their physical location and potential
177 non-uniform growing conditions in the growth chamber.

178 2.2.2 Soil contamination and incubation

179 Prior exposure to plants, soils were spiked with stable Cs and ^{137}Cs . The spiking solution was made by
180 adding 1×10^{-4} mol of stable Cs and 2×10^6 Bq of ^{137}Cs into the minimal solution used to “feed” the plants
181 during the soil exposure step (see below) adjusted to the soil pH. The Cs concentration in the spiking
182 solution was chosen based on the results of Cs sorption isotherm experiments on these soils (data not
183 shown). A thin slice of soil (~ 10 mm) was placed in a beaker, saturated with the minimal solution and
184 contaminated uniformly by dripping the Cs spiking solution onto the whole soil surface. The soil layer was
185 then covered by another thin layer of soil and contaminated in the same way. This protocol was repeated
186 until all the soil and spiking solution were used. At the end of the spiking procedure, contaminated soil
187 reached a ^{137}Cs activity of about 2×10^3 Bq $\text{g}^{-1}_{\text{dw}}$ and a total concentration of added cesium (stable +
188 radioactive) of 1×10^{-7} mol. $\text{g}^{-1}_{\text{dw}}$. Contaminated soil was then incubated for two weeks to allow the
189 establishment of a chemical equilibrium for Cs retention process within the soil (Chaif, 2021; Siroux, 2017).
190 Prior to experiments, contaminated soils were air-dried until a final moisture content of about 30% was
191 achieved, and then well-mixed to homogenize contamination and transferred into the lower part of the
192 Rhizotests. About 20 g of dried soils were placed in 36 Rhizotest lower part devices, corresponding to a 5
193 mm soil layer. Each Rhizotest lower part was connected to a tank containing 800 ml of the minimal
194 solution. Rhizotests were incubated for three days in a growth chamber in the dark at 20°C and 70%
195 relative humidity before experiments. The end of this incubated period was considered as time 0 (day 0)
196 of the experiment.

197 2.2.3 Growing step

198 For each Rhizotest, about 40 mustard seeds and 30 millet seeds were deposited on the Rhizotest upper
199 part to comply with the recommended plant density of the ISO 1698 norm (ISO 16198:2015). 24 Rhizotest
200 upper parts were prepared for each plant. The latter were put on filter paper moistened with $600 \mu\text{mol.L}^{-1}$
201 CaCl_2 and $2 \mu\text{mol.L}^{-1}$ H_3BO_3 to activate germination. Germination was left to start in a growth chamber
202 for 4 days in the dark with a relative humidity of 80%. Seedlings were then transferred to a hydroponic
203 device for three weeks where the hydroponic solution was a full nutritive solution containing $10 \mu\text{mol.L}^{-1}$
204 H_3BO_3 , $2000 \mu\text{mol.L}^{-1}$ $\text{Ca}(\text{NO}_3)_2$, $2000 \mu\text{mol.L}^{-1}$ KNO_3 , $1000 \mu\text{mol.L}^{-1}$ MgSO_4 , $500 \mu\text{mol.L}^{-1}$ KH_2PO_4 , 100
205 $\mu\text{mol.L}^{-1}$ NaFe(III)EDTA , $0.2 \mu\text{mol.L}^{-1}$ CuCl_2 , $2 \mu\text{mol.L}^{-1}$ MnCl_2 , $1 \mu\text{mol.L}^{-1}$ ZnSO_4 , $0.05 \mu\text{mol.L}^{-1}$ Na_2MoO_4 . At
206 the end of the hydroponic step, 21 plants were selected on the basis of their biomass aspect and used for
207 experimentation with contaminated soils.

208 2.2.4 Soil exposure step

209 At the end of the soil incubation time, 18 Rhizotests lower parts were kept bare to be used as unplanted
210 controls and 18 others randomly received their corresponding upper part with pre-grown mustard or
211 millet respectively. Rhizotests were connected to the minimal solution tank used to provide some
212 nutrients to the plants during the test. Its composition ($10 \mu\text{mol.L}^{-1}$ KCl, $50 \mu\text{mol.L}^{-1}$ H_3PO_4 and $750 \mu\text{mol.L}^{-1}$
213 MgSO_4) was reduced to exclude nutrients liable to compete with Cs either for sorption in soils or for
214 uptake by plants. Evapotranspiration was tracked daily during the entire experiment by weighing the
215 minimal solution tank. Any loss in volume was compensated for by adding new solution.

216

217 2.3 Soil and plant sampling and measurements at the end of each exposure period

218 On day 0 (end of preculture/incubation time), 3 upper parts and 3 lower parts were randomly sampled
219 and directly processed. On days 2, 3, 4, 7, 14 and 21, 3 planted Rhizotests and 3 unplanted Rhizotests
220 (except on day 3) were sampled.

221

222 2.3.1 Root and shoot sampling

223 Plants were removed from the Rhizotest and then put into a beaker with 20 mL of minimal solution for 1
224 minute to remove non-adsorbed cesium on the roots and blotted dry on absorbent paper. Then shoots
225 and root mats were sampled separately and air-dried in an oven at 60°C for 1 week until reaching a
226 constant weight. The dry biomass of roots and shoots was digested in a 65% HNO_3 and 30% H_2O_2 mixture
227 at 120°C , then evaporated until dry and dissolved in 20 ml 2% v:v HNO_3 , before analysis.

228 2.3.2 Soil solution sampling

229 During dismantling, the soil solution of each Rhizotest soil was extracted by centrifuging about 15 g_{dw} of
230 contaminated soils at 100 000 g and 20°C for 1 hour (Beckman Avanti J30i). At the end of the
231 centrifugation, supernatant was collected as soil solution, and then filtered with PES 0.8/0.2 μm filter (PALL
232 acrodisc Syringe Filter). The soil pellet was then dried and used to estimate the water content of the soil
233 during the experiment in Rhizotest.

234 2.3.3 Successive extraction experiment

235 Four-stage successive batch extractions were run on each soil type used in the Rhizotests (unplanted or
236 planted soil) at each time except on day 3. About 1 g_{dw} was put into a previously washed dialysis bag

237 (Medicell Membrane Ltd, Size2 inf dia 18/32"-12-14000 Daltons). The dialysis bag was then filled with 10
238 ml of the minimal solution, closed and put into a sealing screw-cap polypropylene centrifugation tube
239 filled with 40 ml of minimal solution to reach a 1/50 soil/solution ratio. Batches were then agitated with
240 an end-over shaker at room temperature. After 24h of agitation (time required to reach a steady state
241 determined from previous study; Chaif, 2021; Siroux, 2017), the dialysis bag containing contaminated soil
242 was removed from the polypropylene tube and put in a new tube with 40 mL of minimal solution for the
243 next 24 h and so on for four times. At each 24h-step, an aliquot of solution was taken to quantify the ¹³⁷Cs
244 desorbed from the contaminated soils.

245 2.3.4 Chemical analyses

246 Major cations in both dry plant biomass and pore water were analyzed by induced coupled plasma-atomic
247 emission spectrometry (ICP-AES, OPTIMA 4300 DV, Perkin Elmer, quantification limit = 10 µg.L⁻¹ for each
248 element). ¹³⁷Cs was measured using a pure germanium gamma spectrometer (Camberra EGPC 42.190.R
249 and GC-3018-7500). All activity measurement were decay-corrected back to the reference date of the
250 source used. Assuming that native Cs was poorly available in these soils, stable Cs concentrations in plants
251 and pore water were calculated from ¹³⁷Cs measurements using the specific activity of the solution used
252 to contaminate the soils.

253

254 2.4 Data analysis

255 2.4.1 Calculating the available fraction of Cs

256 A theoretical model that assumes that Cs in the soil is composed of two pools, one available and in
257 equilibrium with the solution and another one that remains fixed on the solid and does not participate in
258 the soil-solution equilibrium process, was used to interpret the results of the four step batch extraction
259 experiments (Teramage et al., 2018; Coppin et al, in prep). At each step of the extraction, the
260 concentration of Cs sorbed on the soil solid phase ($[Cs]_{solid}$, mol.kg⁻¹) can be expressed as a function of the
261 concentrations of Cs in water ($[Cs]_w$, mol.L⁻¹) and of the Cs fixed on the solid ($[Cs]_{fix}$, mol.kg⁻¹) and
262 modeled based on the following equation:

$$263 \quad [Cs]_{solid} = k_d' \times [Cs]_w + [Cs]_{fix} \quad (1)$$

264 were k_d' ($L.kg^{-1}$) is the partition coefficient of Cs between the available solid pool and the batch solution.
265 By plotting $[Cs]_{solid}$ against $[Cs]_w$ for the 4 steps of the extraction, we can deduce k_d' as the slope of the
266 linear regression and $[Cs]_{fix}$ as the y-intercept.

267 The concentration of Cs in the available solid pool ($[Cs]_{solid_avail}$, $mol.kg^{-1}$) of the soil is deduced from (1)
268 by:

$$269 \quad [Cs]_{solid_avail} = [Cs]_{soil,ti} - [Cs]_{fix} \quad (2)$$

270 where $[Cs]_{soil,ti}$ ($mol.kg^{-1}$) is the total concentration of Cs in the soil used to performed the 4 step batch
271 extraction experiments (corresponding to the concentration of Cs in the soil of the Rhizotest at the time
272 of sampling).

273 The total Cs content that remains available in the Rhizotest at the time of sampling ($C_{S_{soil_avail}}$, mol) was
274 defined as the sum of the available Cs pool and the pore water fraction of Cs. This value is calculated as:

$$275 \quad C_{S_{soil_avail}} = m \times [Cs]_{solid_avail} + V[Cs]_{pw} \quad (3)$$

276 where $[Cs]_{pw}$ ($mol.L^{-1}$) is the concentration of Cs in the pore water of the Rhizotest at the time of sampling,
277 m (kg) the dry mass of soil in the Rhizotest and V (L) the volume of pore water in the Rhizotest soil.

278 2.4.2 Calculating the concentration ratio (C_R)

279 C_R is usually defined as the ratio between the concentration in the plant and the concentration in the bulk
280 soil (which acts as an infinite reservoir). Since, in the Rhizotest, plants may substantially deplete Cs in soils
281 in case of high plant uptake, C_R has been calculated for each experiment using the following equation:

$$282 \quad CR = \frac{[Cs]_{shoot}}{[Cs]_{soil,t=0}} \quad (4)$$

283 Where $[Cs]_{shoot}$ is the concentration ($mol.g^{-1}_{dw}$) of cesium into the shoot and $[Cs]_{soil,t=0}$ is the concentration
284 ($mol.g^{-1}_{dw}$) of cesium in the soil at the beginning of the experiment.

285 2.4.3 Statistical analysis

286 All statistical analyses were performed with R software (R Development Core Team, 2011). Results were
287 subjected to one-way analysis of variance (ANOVA) with Tukey posthoc tests. Absence of auto-correlation
288 was checked by a Durbin-Watson test on residuals. Normality of the distributions and homogeneity of
289 variance were verified by a Shapiro-Wilk and a Levene test respectively. Results of posthoc tests are
290 displayed using different letters. Displayed values are generally the mean results of 3 Rhizotests with their
291 corresponding standard errors (\pm s.e.).

292

293 **3 Results**

294 3.1 Plant growth and evapotranspiration rate in Rhizotests

295 The physiological state of plants in terms of growth conditions and hydric status were assessed through
296 careful assessment of mean evapotranspiration, plant dry biomass and fresh biomass water content.

297 Initial biomass for a given plant was not equivalent for all conditions (Figure S2). However, plant dry
298 biomass globally increased with time in all Rhizotests. For all soil/plant conditions except millet on soil H,
299 this increase mainly concerns the shoots. Finally, in all cases, mustard had a higher biomass than millet.

300 The water content of biomass over the exposure period (Figure S3) was more or less stable for millet roots
301 and shoots for the three soils, except for the shoot for soil E where a 20% decrease was observed after 7
302 days of exposure. The same trend was observed for mustard on soil E, both for shoots and roots, as well
303 as for shoots on soil H. Thus mustard seems to be hydrologically limited on soils E and H and millet partly
304 limited on soil E.

305 In all conditions, dry biomass correlated well with the cumulative amount of water evaporated over the
306 same period (Figure S4) as expected. Mustard exhibited a higher biomass than millet, and thus higher
307 evapotranspiration capacity. It should be noted that the correlation is a bit weaker for mustard on soils H
308 and E, in agreement with the observations recorded regarding the loss of water content in fresh biomass
309 for these two conditions.

310

311 3.2 K concentration in soil solution

312 The K concentration in the soil solution of each Rhizotest is presented on Figure 1. Due to the very low
313 water content of soil S, soil solution extraction was often too limited to measure K concentrations,
314 particularly for millet.

315 In unplanted Rhizotests, the K concentration remained nearly constant over the time-frame of the
316 experiments, except a small decrease with time in the experiment with soil H conducted in parallel to the
317 millet experiment. The three soils exhibited different K concentrations in soil solution, with values above
318 or below the threshold of 250-300 $\mu\text{mol.L}^{-1}$ that drives the involvement of the different K^+ transport
319 pathways in plants (Zhu and Smolders, 2000). The K concentration in the soil S solution was always above
320 this threshold, around 1 mmol.L^{-1} , suggesting that Cs uptake through the K^+ channel dominates. On the

321 other hand, the K concentration in soil H is the lowest value of the three soils, at around $100 \mu\text{mol.L}^{-1}$,
322 indicating that Cs transfer could occur mainly through K^+ transporters. The K concentration in soil E was
323 intermediate, around $200 - 500 \mu\text{mol.L}^{-1}$.

324 Generally, plants had no effect on the concentration of K in soil solutions; except for experiments with
325 mustard on soil S where a depletion of the concentration of K in soil solution of more than 1 order of
326 magnitude was recorded compared to controls after 4 days of exposure to plants.

327

328 3.3 K in the plants

329 K results are displayed on Figure 2 and in supplementary material - concentrations in Figure S5 and
330 concentration ratio in the shoots and in the roots in Figure S6. Considering plant biomass dynamics, these
331 concentrations led to more than 75% of the total K in plants being in the shoots in all conditions (except
332 on day 21 for millet, for which this value decreased to 50%) (Figure 2). K stock in plants was more or less
333 stable in all experiments, except in soil E. After 14 days of exposure, K stock tended to decrease, however
334 it is only significant for millet on day 21. This corresponds to a K concentration which falls from values
335 between 0.5 and 2 mmol.g^{-1} for both plants at the beginning of the experiment to a final concentration
336 around ten times lower. Variation in K shoot-to-K root concentration ratios over time was quite similar for
337 both plants on soil E (Figure S6). Ratios were also similar for both plants on each sampling date except day
338 21 where the mustard ratio was higher than the millet value. For the 2 other soils, millet exhibited higher
339 ratios whatever the sampling date for soil S and after 7 days for soil E.

340

341 3.4 Concentration of Cs in soil solution and availability of Cs in soils

342 The Cs concentrations in the soil solution of each Rhizotest is presented on Figure 1. Similarly to K, it was
343 sometimes impossible to extract enough solution from soil S to measure Cs concentrations. Marked
344 differences appeared between soils; with values covering a range of three orders of magnitude.

345 Similarly to K, the Cs concentration in soil solution remained stable in unplanted Rhizotests during the
346 whole experiment; suggesting that the soil and solution reached a steady state. Soil E exhibited a very
347 small amount of Cs in soil solution (around $0.01 \mu\text{mol L}^{-1}$), whereas the Cs concentration in soil solution
348 reached $1 \mu\text{mol.L}^{-1}$ in soil H and nearly $10 \mu\text{mol.L}^{-1}$ in soil S. This reflects the difference in Cs availability
349 (Figure S7), assessed through the desorption method described in section 2.4.1. Cs available fractions in
350 soils E and H are low (around 1-10% of the Cs added to soils) and contrastingly higher for soil S ($35 \pm 10\%$).

351 Contrary to K, the effects of plant uptake on variation in Cs concentration in soil solution over time could
352 be observed for mustard in soils H and S. In the same way as K, the decrease was sharpest during the four
353 first days of experiments, and then the Cs concentration in soil solution stabilized until the end of the
354 experiment, suggesting a new steady state between soil and solution. The final Cs concentration in soil
355 solution was lower, from 1 to 2 orders of magnitude compared to controls. The quantity of Cs in the fixed
356 fraction of soil S for millet and mustard and soil E for mustard seemed to decrease with time (Figure 3).
357 However, we could not confirm this finding due to the large uncertainty for the calculated fixed Cs pool.

358

359 3.5 Cs uptake by plants

360 Cs results are displayed in Figure 3 in terms of Cs distribution within the different Rhizotest compartments
361 (plant, fixed and available fraction in soil), in Figure 4 for the root/shoot distribution of Cs stock and Figure
362 5 for calculated CRs. As a complement, Cs plant uptake rates, plant Cs concentrations and Cs concentration
363 ratios in the shoots and roots are shown as supplementary material in Figures S6, S8 and S9 respectively.

364 The quantity of Cs taken up increased continuously with time over the 21 days of exposure, for both plants
365 and for the three soils. However, the maximal quantities accumulated differed between plants and soils.
366 Both plants accumulated more Cs when grown on soil S (about 30-35% of the initial Cs added to the soils)
367 than on soils E and H, for which the accumulated quantities stayed between 1-5% and 8% of the initial Cs
368 added to the soils, respectively. Regarding plants, mustard accumulated more Cs than millet after 21 days
369 (3 to four times more, when expressed in moles), except when grown on soil H. Yet the uptake rate of Cs
370 decreased with time for all soil/plant combinations (Figure S8).

371 The distribution between roots and shoots depends on the type of plants and the duration of the
372 experiments. Over short exposure periods, roots were the main compartment for Cs (except for millet on
373 soil S), however accumulation in shoots prevailed over longer exposure periods. The distribution for
374 mustard varied little for all soils, with a distribution of about 40% of Cs in roots and 60% in shoots after 21
375 days of exposure (Figure 4). The same trend was recorded for millet grown on soil S. However, when grown
376 on soils E and H, roots remained the main compartment for Cs accumulation in millet. Shoot-to-root Cs
377 concentration ratio showed the same trend with time for all soils for mustard, which is an increase at later
378 sampling dates. This time trend was also recorded for millet on soil E with limited intensity, and on soil H
379 with higher intensity (Figure S6). On the other hand, higher values are recorded for soil S at earlier
380 sampling dates. A contrasted translocation between plants is recorded for the earlier sampling dates for

381 soil S (millet > mustard) or later sampling dates for the other 2 soils (E and H). In addition, these 2 soils
382 contrast as mustard records a higher translocation than millet on soil E, but a lower translocation on soil
383 H.

384 The CR of Cs (Figure 5) globally increased with exposure time for both plants and the three soils, which
385 reflects the dynamics of element uptake by plants driven by growth during the experimental period. The
386 magnitude of this increase depended on both soil and plant types. After 21 days of exposure, the CR of
387 plants grown on soils E and S were about 3 to 6 times higher than CR after 2 days of exposure; whereas on
388 soil H, CRs remained nearly the same during the whole experiment. Whatever the exposure time, the three
389 soils could be distinguished by the absolute values of CR. Whereas CR values for soil E are more or less
390 equivalent to values obtained for soil H for the two plants, CRs for soil S were 230 and 36 times superior
391 to values for soil E for millet and mustard respectively, after 21 days.

392 Regarding the hypothesis and associated experimental choices defining this study, Cs accumulation in both
393 plants was, as expected, higher for soil S than for soils E/H, with differences between soil E and H that
394 have to be addressed further in the discussion section. However, the relative accumulation behaviour of
395 Cs between mustard and millet depends on soil types, with millet unexpectedly exhibiting a higher CR than
396 mustard in soil S, which shows that soil/plant interactions shape the plant response in a given soil.

397

398 **4 Discussion**

399 Although a large amount of literature has highlighted the role of the different processes involved in Cs
400 uptake by plants, there is still no simple way to predict the transfer of Cs for a specific plant from a
401 particular soil. Thus we defined our experimental plan assuming a constant relative CR between plant
402 species for all soils as proposed by the phylogenetic analysis methodology (Willey, 2010) and the concept
403 of a “benchmark-taxon” (Beresford and Willey, 2019). Our experimental plan devoted to soil-to-plant
404 transfer of Cs was applied to a limited number of plant/soil combinations but related to this global
405 framework: two plants, millet and mustard belonging to the Poales and Brassicales order respectively
406 (from the Monocot and Eudicot clades respectively) in three soils with different physico-chemical
407 properties (pH, clay and organic matter content) known to impact Cs sorption and availability in soils.
408 Experiments performed in Rhizotests led to unexpected results, such as the ranking of recorded CRs for
409 the 2 plants ($CR_{\text{mustard}} > CR_{\text{millet}}$ for soil E, no differences in CR for soil H and $CR_{\text{millet}} > CR_{\text{mustard}}$ for
410 soil S) do not conform to the global framework. To “explain” this discrepancy, we could firstly refer to the

411 limits of the methodology used and secondly to the possible parameters responsible for differences
412 recorded (soil effect, plant parameters like physiology, root uptake, root-shoot translocation, etc.) without
413 occulting the limits of the experimental set-up.

414
415 There are a limited number of studies and thus CR values of Cs acquired in the Rhizotest device available
416 for the purpose of comparisons (Guivarch et al., 1999; Staunton et al., 2003; Cherif, 2017). Data provided
417 in Staunton et al. (2003) can particularly be used to compare results for close plant species belonging to
418 the same family (rape vs mustard, brome vs millet) and 2 type of soils sandy (S) and clayey (E). The CRs of
419 mustard and millet match the values of homologous plants for clayey soils but are considerably higher for
420 sandy soils. Unlike our results, the ranking of CRs was the same for the two plants for all soils, with brome
421 having the lowest CR. Indeed, Brassicales order tend to absorb more cesium than the Poales order (Frissel
422 et al., 2002; White et al., 2003; Willey et al., 2005; Willey, 2010; Ogura et al., 2014).

423 Looking at the dataset of Willey et al. (2005), some species in a given order (millet in this case) may exhibit
424 a contrasting behaviour compared to most other species in the same order. In addition, datasets are
425 sometimes unbalanced regarding soil types, Cs availability, plant taxa diversity, study types or steady-state
426 conditions. For example, only 6 data are available for millet (*Panicum Millaceum*) from the 972 data used
427 by Beresford and Willey (2019) to derive REMLmean. Thus, as a result, the ranking between two species
428 may not always be correctly assessed, and may be updated if additional data is added to the dataset.
429 Consequences can be bad CR predictions if that particular species is chosen as the reference one in the
430 “benchmark-taxon” approach as recently discussed in Beresford et al. (2020). Besides, REML analysis
431 considers plant species as the fix factor and “study” (aggregating, soil, availability, experimental
432 conditions...) as random factor. Yet interaction problems between these two factors are highly probable.

433
434 Cs uptake by plants depends on different processes that interact together: the soil offer, the buffer
435 capacity, the exploitation extent of the offer by the roots and plant needs. The soil offer, or environmental
436 availability, corresponds to the level of Cs in soil solution and in the available solid pool. The buffer effect
437 is the capacity of the solid phase to feed the soil solution when it is depleted. Clay content, CEC, and
438 organic matter are physico-chemical parameters known to play a role in Cs retention and soil availability
439 (Burger and Lichtscheidl, 2018). The Cs concentration in soil solution (Figure 1) and fraction of available Cs
440 (Figure 2) increased in the order soil E < soil H < soil S. This order exactly matches potential predictions

441 based on the clay content and CEC of the three soils. The highest the clay content and the CEC, the lower
442 Cs availability. When explaining Cs retention in soils, the nature of the clays is also important. Clays in both
443 soils E and H mainly consist of illite in this case, a mineral known for its high capacity to sorb Cs (Cherif et
444 al., 2017). It should be noted that the higher level of organic matter and the lower pH of soil H (which
445 could have favoured more Cs retention in this soil) did not compensate the lower clay content and CEC
446 when compared to soil E.

447 When comparing Cs in plants to the pool of Cs available in soils, it seems that for soil E mustard was able
448 to take up more Cs than the quantity present in the pool available at the beginning of the experiment
449 (Figure 2). For soil S, the pool of available Cs seems to have been re-supplied by the soil “fixed” pool of Cs
450 during the timeframe of experiments. This finding must be considered with caution as uncertainty is high
451 for the quantification of the available and fixed pools. Yet it is true that, with the Rhizotest design, the
452 small volume of soil is submitted to high root activity, through exudation for example, that can favour Cs
453 desorption through mechanisms such as the modification of the cationic exchange capacity of the soil as
454 recorded in Guivarch et al. (1999). In addition, as demonstrated by Teramage et al. (2018) the
455 quantification of available and fixed pools of Cs in soils with the successive 4-step desorption method is
456 highly affected by the nature of the extractant (ammonium acetate predicts less fixed Cs than water).
457 Thus, with the protocol used, Cs available pools for plant uptake could have been underestimated in
458 comparison with desorption driven by root exudates.

459 A lot of studies have unsuccessfully tried to correlate CR with soil physico-chemical parameters (Smolders
460 et al., 1997; Frissel et al., 2002; Nisbet and Woodman, 2000; Massas et al., 2002). Assuming that the plant
461 uptake of Cs correlates directly with Cs soil availability allows us to define a new transfer factor based on
462 the available fraction of Cs instead of the total concentration of Cs ($CR_{avail} = \frac{[Cs]_{shoot}}{[Cs]_{soil_avail,t=0}}$, where
463 $[Cs]_{shoot}$ is the concentration (mol.g^{-1}_{dw}) of cesium into the shoot and $[Cs]_{soil_avail,t=0}$ is the
464 concentration (mol.g^{-1}_{dw}) of cesium available in the soil at the beginning of the experiment). When
465 compared to CR, using CR_{avail} reduced the difference in Cs uptake capacity of plants due to the different
466 nature of soils (Figure 6). This is especially true for mustard, for which a unique value of CR_{avail} may depict
467 its capacity to take Cs up for soils E and S after 21 days. For soil H, this value is a bit less than 10 times
468 lower. No single value may depict millet’s Cs uptake capacity for the three soils. However, the difference
469 between the highest and the lowest CR_{avail} for millet is only a factor of 20 (compared to 200 for CR). CR_{avail}
470 is determined based on simple and easy experiments. Compared to more sophisticated plant transfer
471 models, such as the Absalom model, it seems a promising compromise that could help to reduce the

472 variability of transfer factors for a given plant without any need to deeper characterise the soil or the
473 chemical composition of the soil solution.

474
475 Although our experimental conditions differed from the usual set-up with RHIZOtest designs (longer
476 exposure time and use of a minimal nutritive solution), biomass increase (similar for different soils, Figure
477 S2) and the absence of visible physiological stress signs (like chlorosis or wilting) show that the Rhizotest
478 design was adequate for plant growth. The use of a root mat constraints the geometry of roots in soil, thus
479 removing one of the parameters that can explain differences between species as recorded by phylogeny.
480 In addition, at the beginning of the experiment, as the membrane area was fully covered with roots (= the
481 effective area of root-soil exchange), the highest uptake capacity is reached and stabilized. Consequently,
482 differences in root morphology or growth cannot account for the recorded differences.

483 A different efficiency of water use for biomass production (amount of dry matter produced per mL of
484 evaporated water) was recorded between the plants. This value remained constant for the three soils
485 (around $0.0018 \text{ g}_{\text{dw}} \cdot \text{mL}^{-1}$) for mustard, however it differed strongly between the soils for millet. As a species
486 adapted to harsh environments (Amadou et al., 2013; Habiyaemye et al., 2017), millet is expected to have
487 lower water needs than mustard or higher water use efficiency (Adak et al., 2013; Nielsen and Vigil, 2017).
488 This was the case for soil E, but not soils H and S where the efficiency of water use of millet decreased by
489 a factor of around 5. Yet, these differences did not alter the efficiency of Cs uptake, which is highest for
490 soil S.

491
492 Since plant Cs uptake occurs through K-uptake pathways, any change in Cs accumulation may be related
493 to changes in K uptake and use in plants.

494 Plant Cs stocks increased with contact time in both roots and shoots, meaning that conditions for uptake
495 and translocation are stable throughout the experiments and that decreases, in particular after 14 days,
496 of water fluxes, water status or K concentrations in plant, which are all interrelated processes (Osakabe et
497 al., 2013; Smith et al., 2019), probably did not result in a significant impact on the Cs uptake capacities of
498 plants.

499 Whatever the conditions (plant, soil, time), the ratio between K and Cs concentrations within the soil
500 solution was more than 10^3 which means that competition between the 2 ions *per se* has been negligible.
501 As shown on Figure 1, in soil E and H, K concentrations in soil solution stay, most of the time, within a

502 range where Cs/K-discriminating K channels (like AtAKT1 in *Arabidopsis thaliana*) are active. The threshold
503 below which high-affinity transporters (like HAK5 and their homologs in other plant species, Qi et al., 2008;
504 Nieves-Cordones et al., 2017, 2020) co-exist with channels seems to depend on species ($250\text{-}350\ \mu\text{mol.L}^{-1}$
505 K) and is not precisely known for the test species. Yet a shift to high-affinity transporters results in an
506 increase in flux (Genies et al., 2017; Nieves-Cordones et al., 2020). However, despite the decrease in K
507 with time down to low range recorded for H and soil S for mustard in particular, the root uptake was not
508 substantially affected.

509 Root-to-shoot translocation of K or Cs may also be species-specific and some authors like Staunton et al.
510 (2003) suggested that differences in Cs uptake may be more due to differences in translocation capacities
511 than soil parameters or root uptake parameters. Mustard, through its higher CR, is supposed to have a
512 higher overall Cs accumulation capacity than millet, and also better translocation as CR is calculated based
513 on shoot concentration. However, as shown by the shoot content/root content ratio for Cs, this is true
514 only for soil E and millet seems to have a better translocation capacity in our conditions, for soil S for the
515 earlier period and soil H for the later period. Conclusions are the same regarding K translocation. Thus,
516 differences in translocation capacity, seem to “explain” the differential behaviour of the 2 plant species
517 on the 3 soils.

518

519 **5 Conclusion**

520 CR values derived from Rhizotest soil-to-plant Cs transfer experiments with millet and mustard and three
521 soils over 21 days highlighted strong variability among soils and plants. For millet, CR varied by two orders
522 of magnitude between the three soils, whereas variation in CR for mustard was only one order of
523 magnitude. More interestingly, millet behaved differently to mustard as a function of soils, with the CR of
524 mustard higher than the CR of millet for soil E, equal for soil H and lower for soil S, in apparent
525 contradiction with the framework underlying the experimental plan design.

526 Accounting for Cs soil availability, and defining a new CR based on the amount of Cs available in the soil
527 (CR_{avail}) can be used to decrease the range of variation in CR for a given plant between the soils, by one
528 order of magnitude. In our study the amount of Cs available was determined through a successive 4-step
529 batch desorption method, which is easily applicable and not time-consuming. Other methods may also
530 exist that could be compared to this one. As an interesting alternative to the use of CR, the robustness of
531 this approach should be further tested on different plants and soils.

532 Regarding the relative behaviour of millet compared to mustard as a function of soils, accounting for Cs
533 availability in soils was not sufficient to explain our results. Different parameters linked to plant physiology,
534 including growth, water use efficiency and K uptake and distribution in plants in relation to K level in soil
535 solution, also failed also to explain the behaviour of millet on soil S. Differences in translocation to shoot
536 is the only parameter that seems to partly “explain” the differences recorded, for both Cs and K and the
537 unexpected behaviour of millet on soil S in particular. This could be a particularity of millet, a species
538 adapted to arid environments and light soils, such as soil S. This highlights the need for further research
539 to better understand the functioning of plants – more specifically intra-taxon - in different soils.

540 Given the low number of plants/soils tested, our results do not lead to reconsideration of the methodology
541 used to derive CR values based on phylogeny. However, they certainly highlight the need to increase the
542 numbers of soils considered for a given plant in the databases that support the establishment of such an
543 approach.

544

545

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550

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744

745 *Table 1: Physicochemical characteristics of the soils used.*

	Soil-E	Soil-H	Soil-S
Clay (%)	18.2	13.1	3.1
Silt (%)	47.3	54.1	0.7
Sand (%)	34.5	32.8	96.2
pH (H ₂ O)	7.5	5.5	9.3
Organic matter (g/kg)	20.3	49.4	1.4
N (g/kg)	1.1	2.6	0.04
Total CaCO ₃ (g/kg)	49	14	118
Exchangeable cations (cmol+/kg)			
CEC	9.89	7.64	1.11
K ⁺	0.383	0.321	0.228
Na ⁺	0.062	0.202	0.139*
Ca ²⁺	34.3	3.23	31.2
Mg ²⁺	0.466	0.598	0.715
Mineralogy**			
CEC <2µm (cmol+/kg)	49.7	20.0	18.7
Illite (%)	33.5	37.9	1
Montmorillonite (%)	25.25	11.11	20
Kaolinite (%)	10.3	20	6.5

746 *exchangeable Na⁺ in soil S was measured by cobaltihexamine extraction

747 **analyses performed on the soil fraction below 2 µm

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749

750 **Figure Captions**

751

752 Figure 1: Potassium (dot) and cesium (triangle) pore water concentration (n=3). Filled dots represent the
753 unplanted Rhizotest data experiment. Empty dots represent the planted Rhizotest experiment. A log
754 scale has been used on the y-axis

755 Figure 2: Potassium stock in the plant and its distribution. The black fraction represents potassium in the
756 shoot, the white fraction represents potassium in the root. K stock in the plant without a common letter
757 differs significantly ($p < 0.05$). If no letters are given, statistics failed to provide evidence of any
758 differences between samples.

759 Figure 3: Variation in cesium quantities, expressed as a percentage of the initial cesium added to the soil,
760 in the plants (in black), in the available fraction (in white) and in the fixed fraction (hatched) of the soils
761 during the Rhizotest experiment. The line represents the Cs fixed fraction at time 0 and the hatched
762 line the uncertainty for this value.

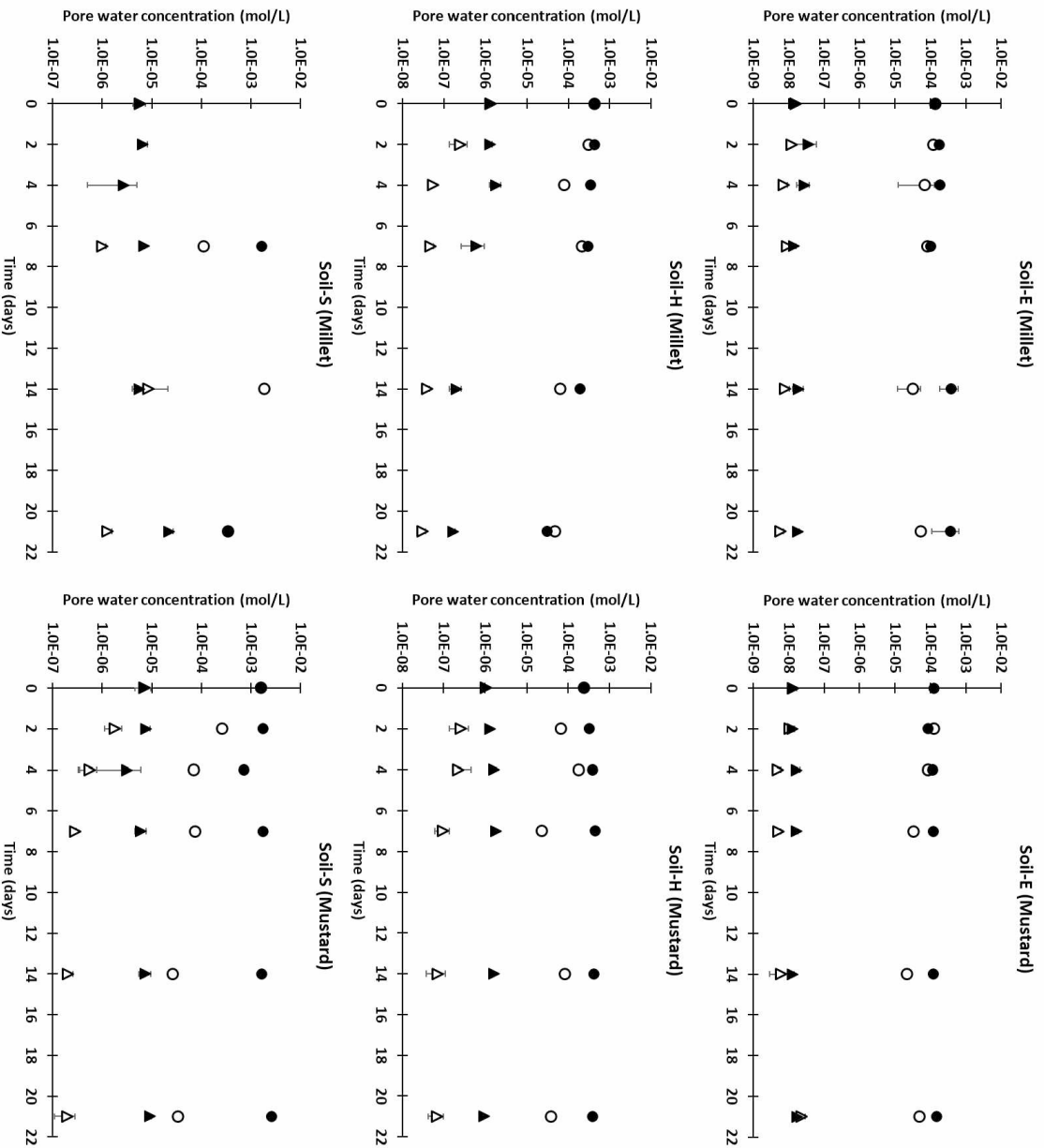
763 Figure 4: Cesium stock in the plant and its distribution. The black fraction represents cesium in the shoot,
764 the white fraction represent cesium in the root. The Cs stock in the plant without a common letter
765 differ significantly ($p < 0.05$).

766 Figure 5: The Cs concentration ratio for all experiments. The concentration ratios for mustard are
767 represented by filled dots, the concentration ratios for millet are represented by empty dots.

768 Figure 6: The Cs concentration ratios based on the available Cs pool in soils for all experiments. The
769 concentration ratios for mustard are represented by filled dots, the concentration ratios for millet are
770 represented by empty dot.

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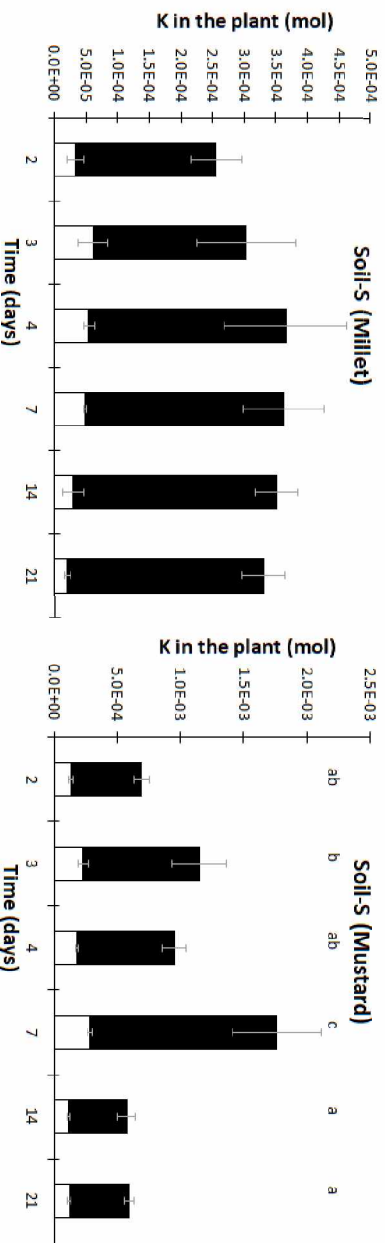
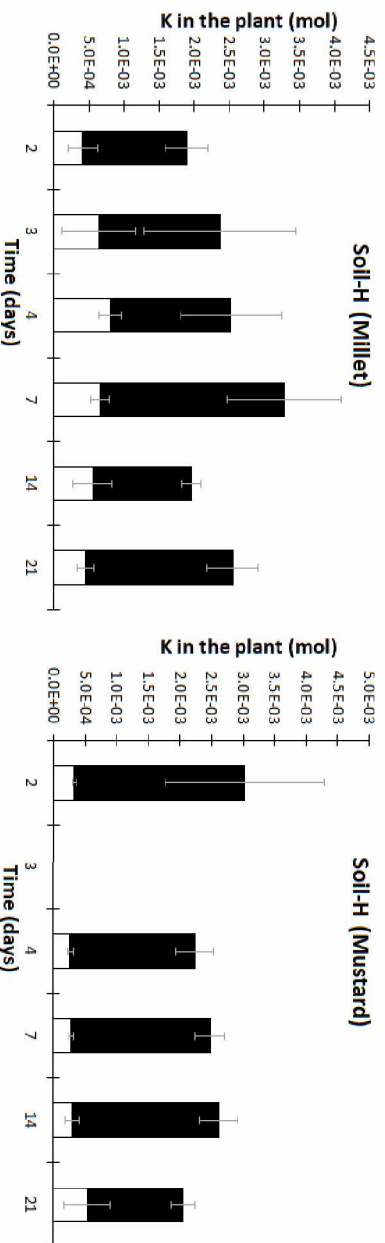
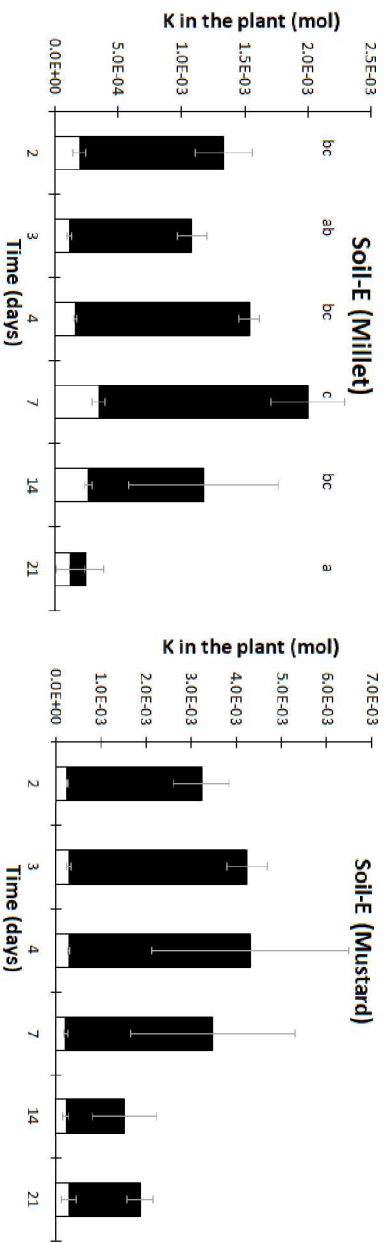
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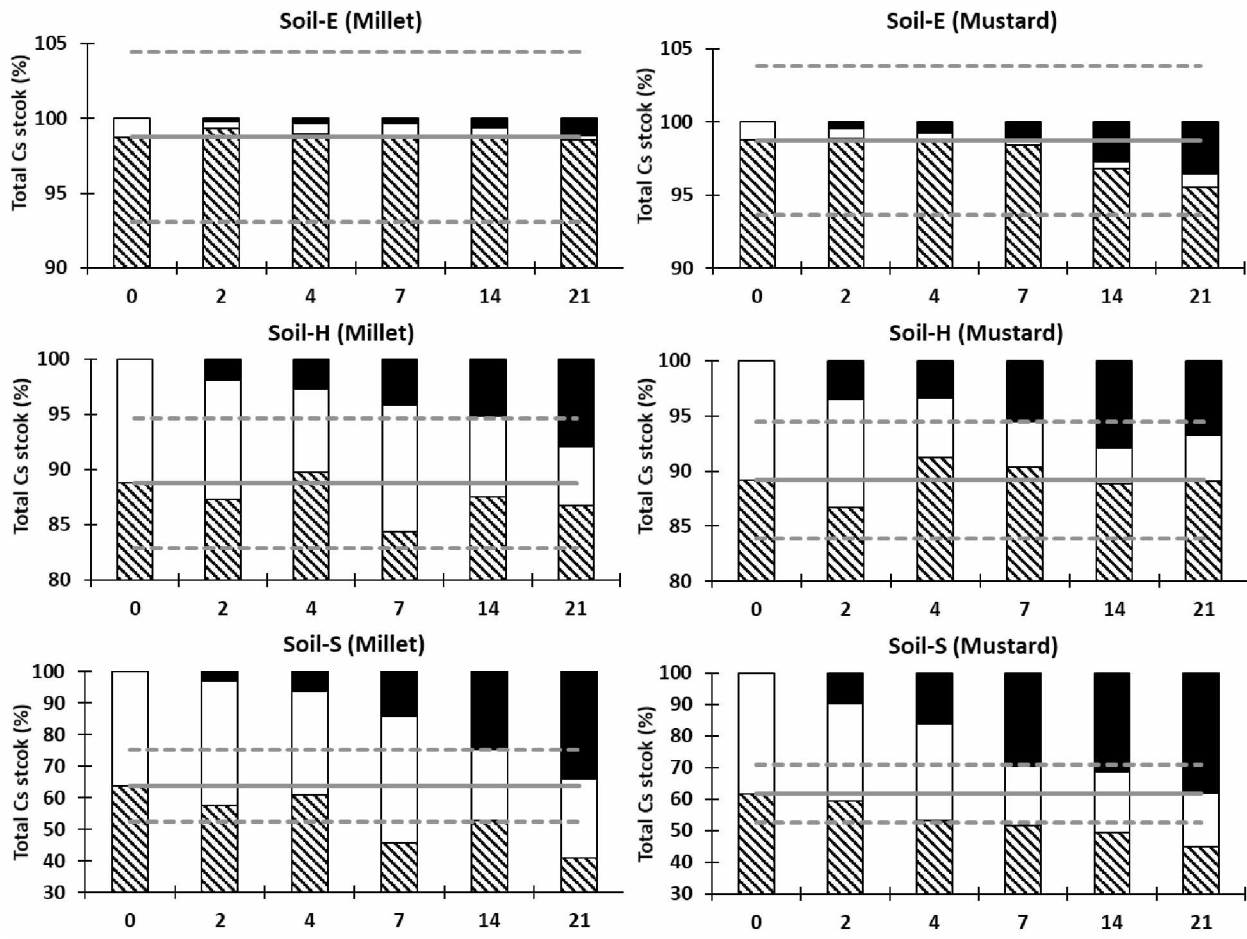
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774 Figure 1

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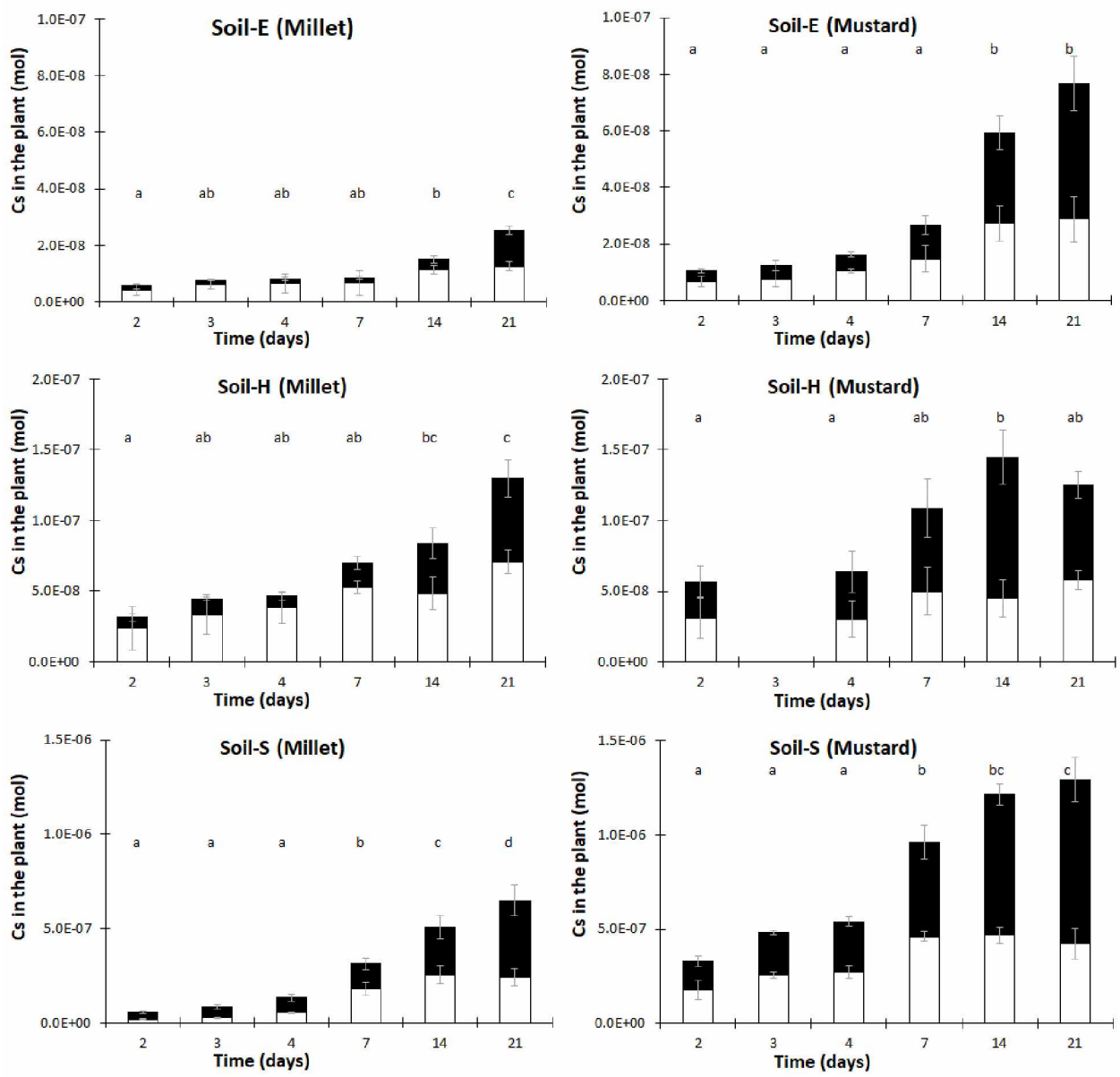
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778 Figure 2



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781 Figure 3

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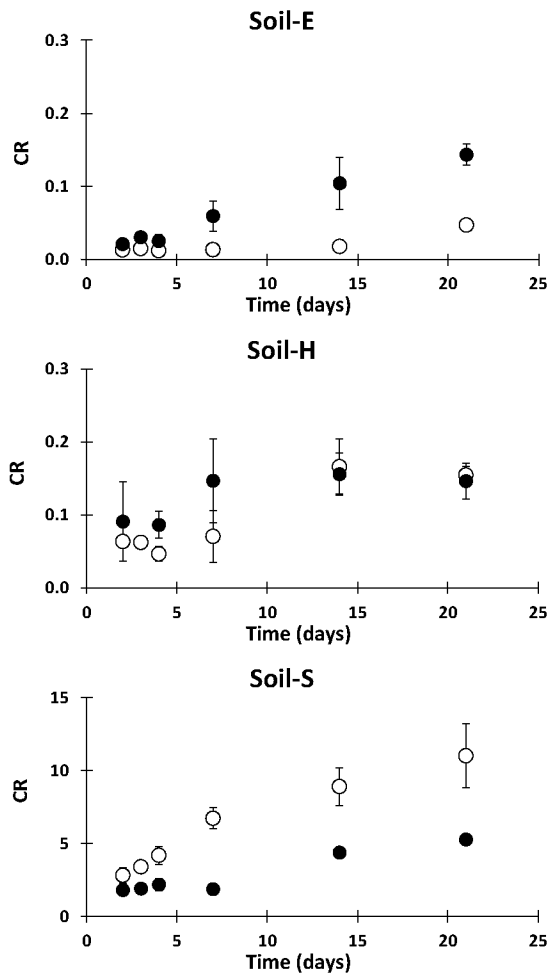


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785 Figure 4

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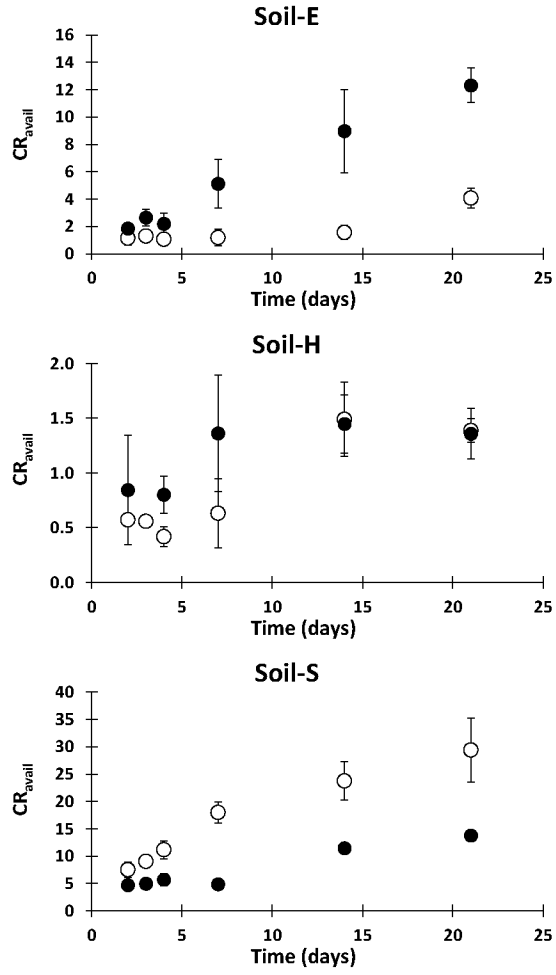


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789 Figure 5

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793 Figure 6

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Supplementary data

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800 **Figure Captions**

801

802 Figure S1a: RHIZOtest device scheme.

803 Figure S1-b: Experimental set-up of one experiment (defined by a soil/plant combination).

804 Figure S2: Variation in average biomass in the RHIZOtest during the experiment. White dots represent root
805 dry-weight biomass, black dots represent shoot dry-weight biomass and grey dots represent the total
806 dry-weight biomass of the plant.

807 Figure S3: Variation in the water content of the biomass in the Rhizotests during the experiments. Empty
808 dots represent the water content of the roots, filled dots represent the water content of the shoots

809 Figure S4: Relationship between biomass and cumulative water evaporated in the planted Rhizotests
810 during the experiment

811 Figure S5: Concentration of potassium in the plant and its distribution (white: potassium concentration in
812 the root, black: potassium concentration in the shoot, hatched: potassium concentration in the whole
813 plant)

814 Figure S6: Concentration ratio in the shoot versus the root for cesium (left) and potassium (right) (white:
815 millet; black: mustard)

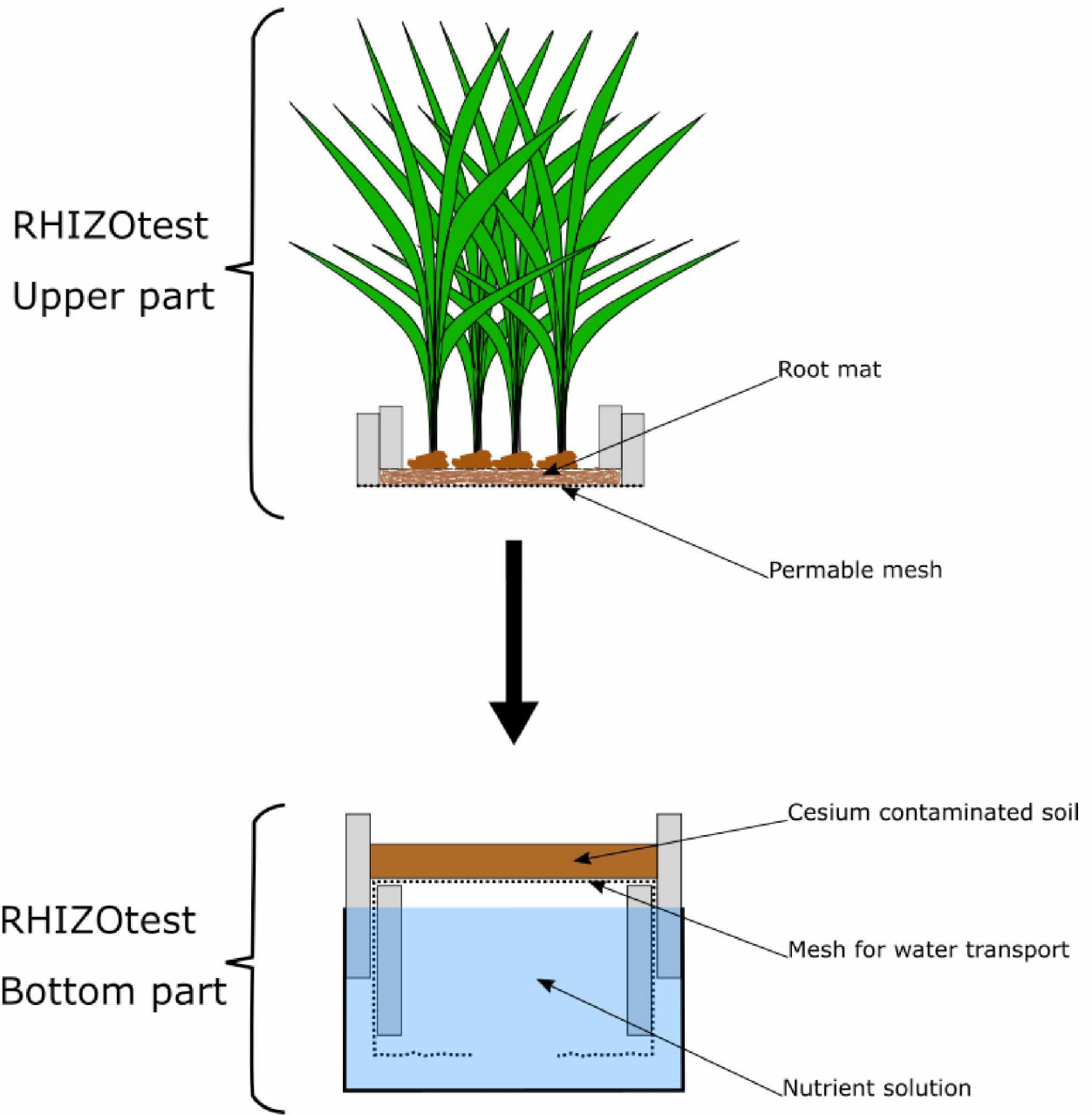
816 Figure S7: Variation in cesium quantities, expressed as a percentage of the initial cesium added to the soil,
817 in the available fraction (white) and in the fixed fraction (hatched) of the soils during the experiment in
818 unplanted Rhizotests

819 Figure S8: Cesium plant uptake rate for each experiment. Empty dots represent the millet experiment and
820 filled dots represent the mustard experiment

821 Figure S9: Concentration of cesium in the plant (white: cesium concentration in the root; black: cesium
822 concentration in the shoot; hatched: cesium concentration in the whole plant)

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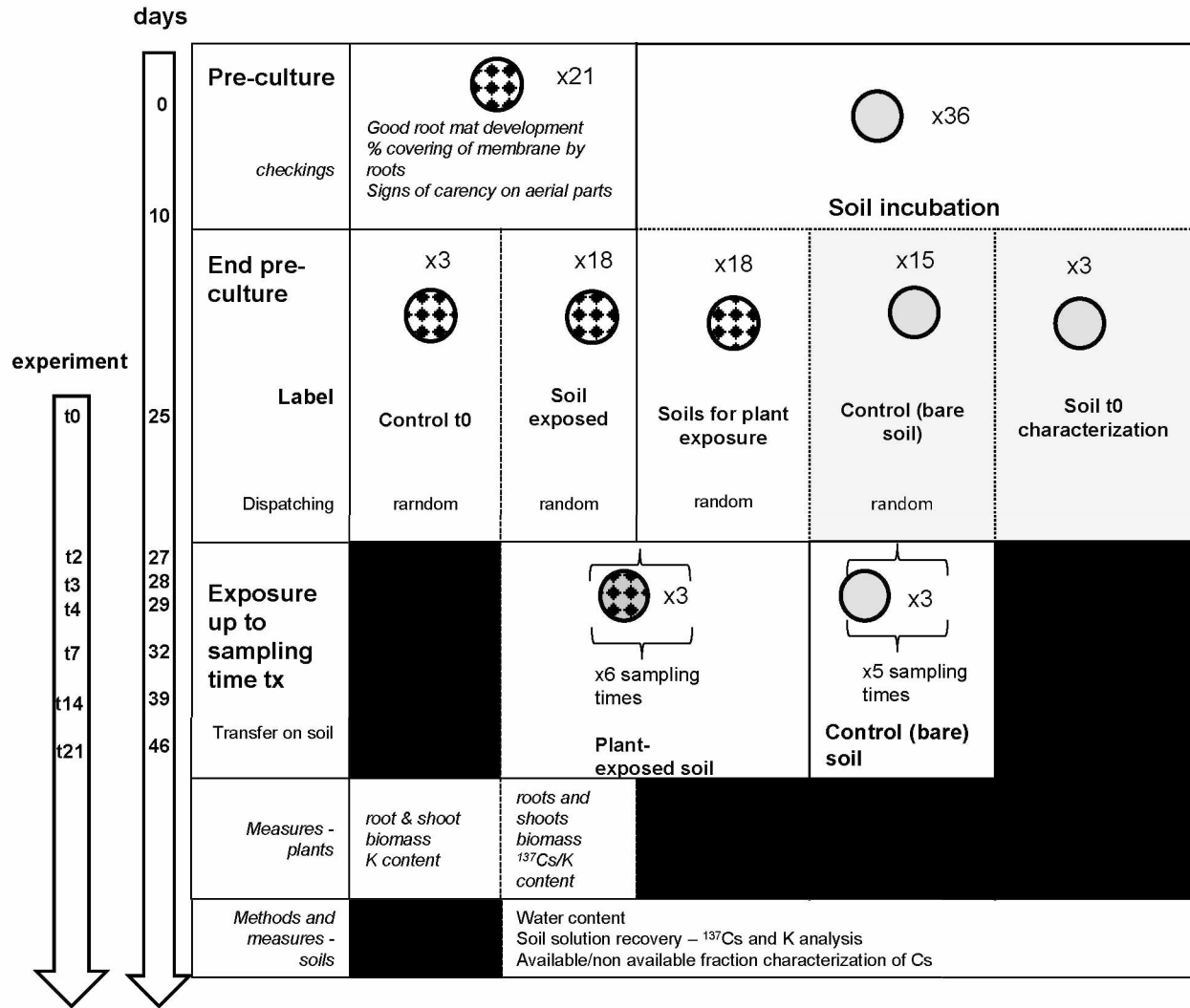
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827 *Figure S1-a: RHIZOtest device scheme.*

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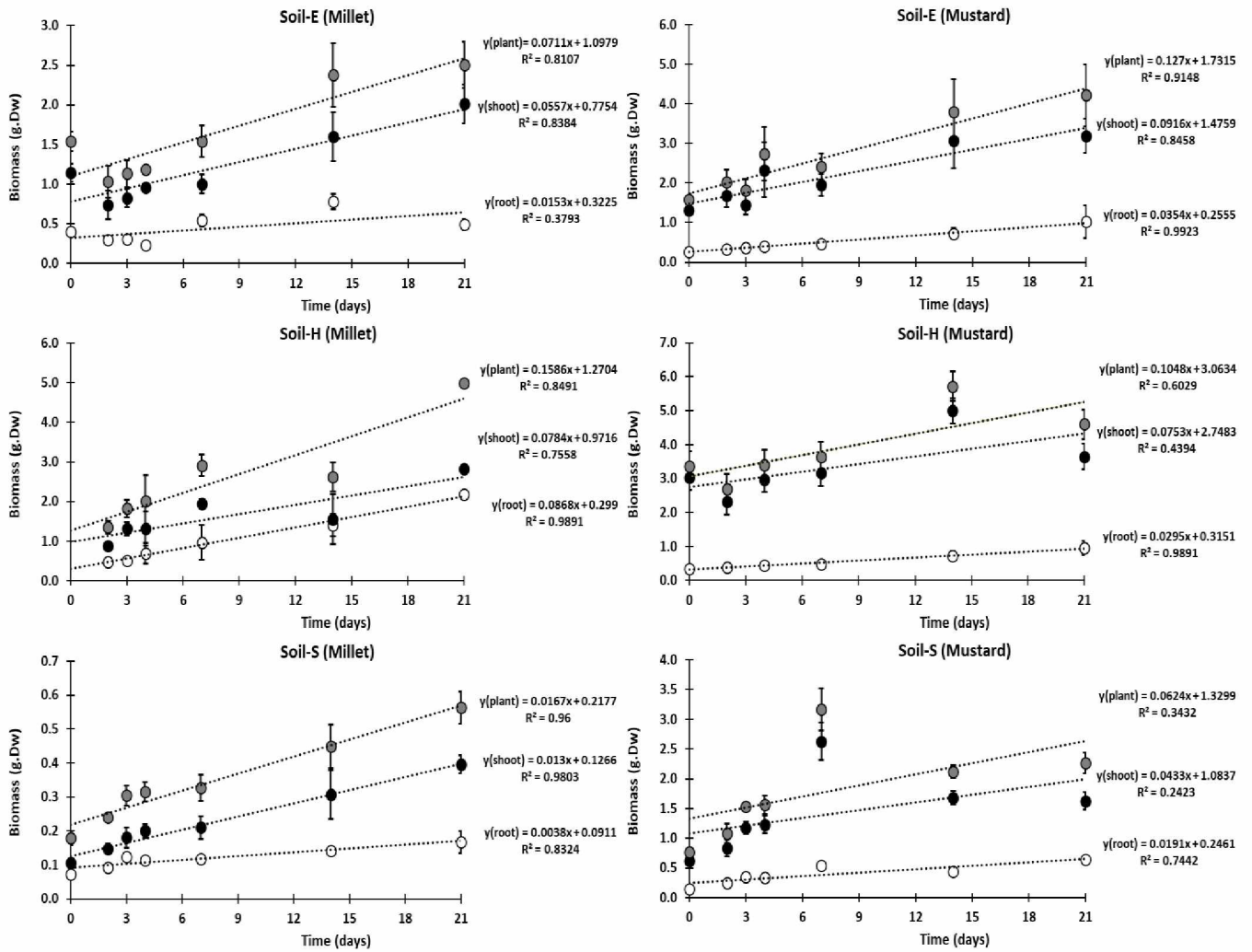
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830 *Figure S1-b: Experimental set-up of one experiment (defined by a soil/plant combination).*

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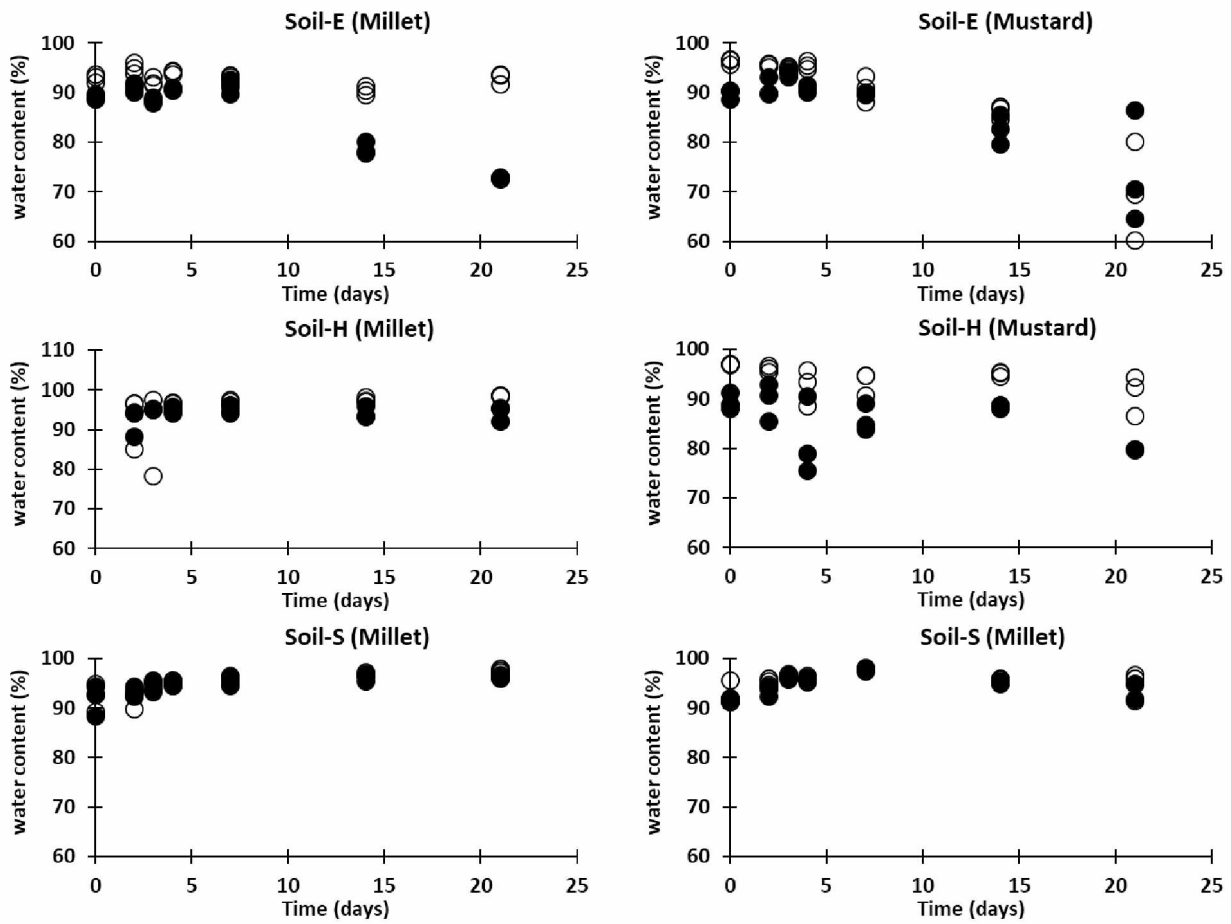
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835 *Figure S2: Variation in average biomass in the RHIZOtest during the experiment. White dots represent root*
 836 *dry-weight biomass, black dots represent shoot dry-weight biomass and grey dots represent total dry-*
 837 *weight biomass of the plant.*

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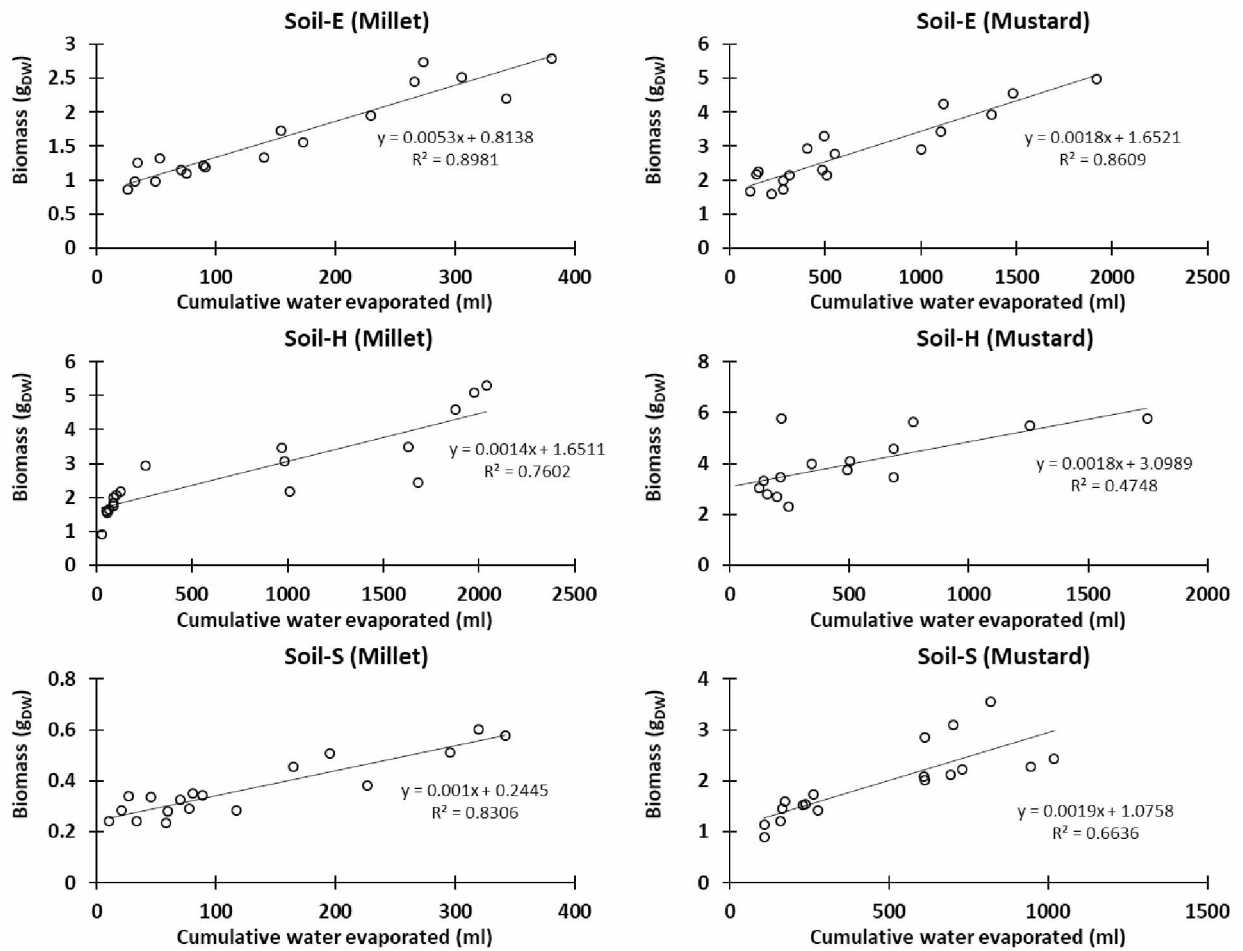


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841 *Figure S3: Variation in the water content of the biomass in the Rhizotests during the experiments. White*
 842 *dots represent water content in the roots, black dots represent water content in the shoots*

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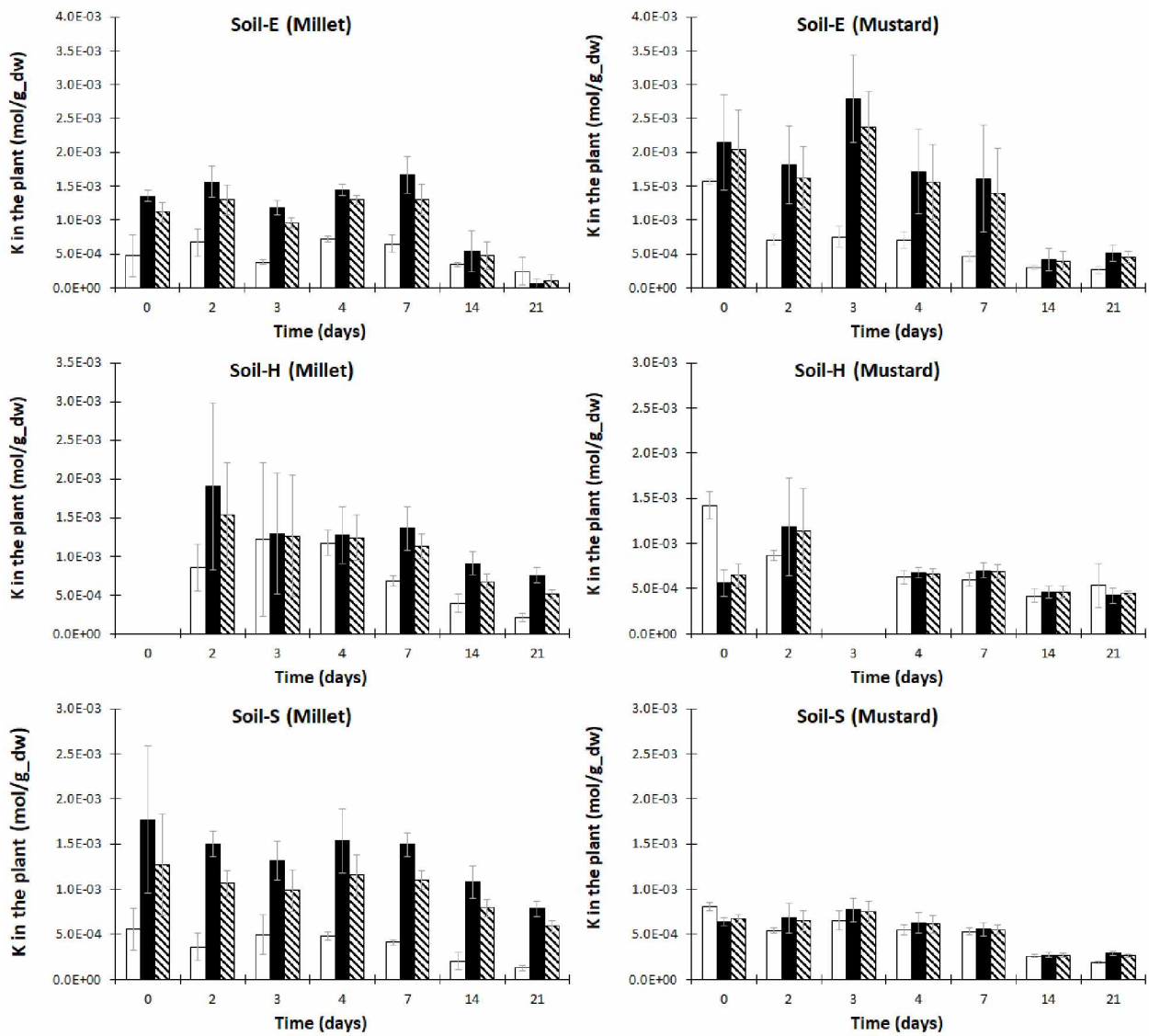
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846 *Figure S4: Relationship between biomass and cumulative water evaporated in the planted Rhizotests*
847 *during the experiment*

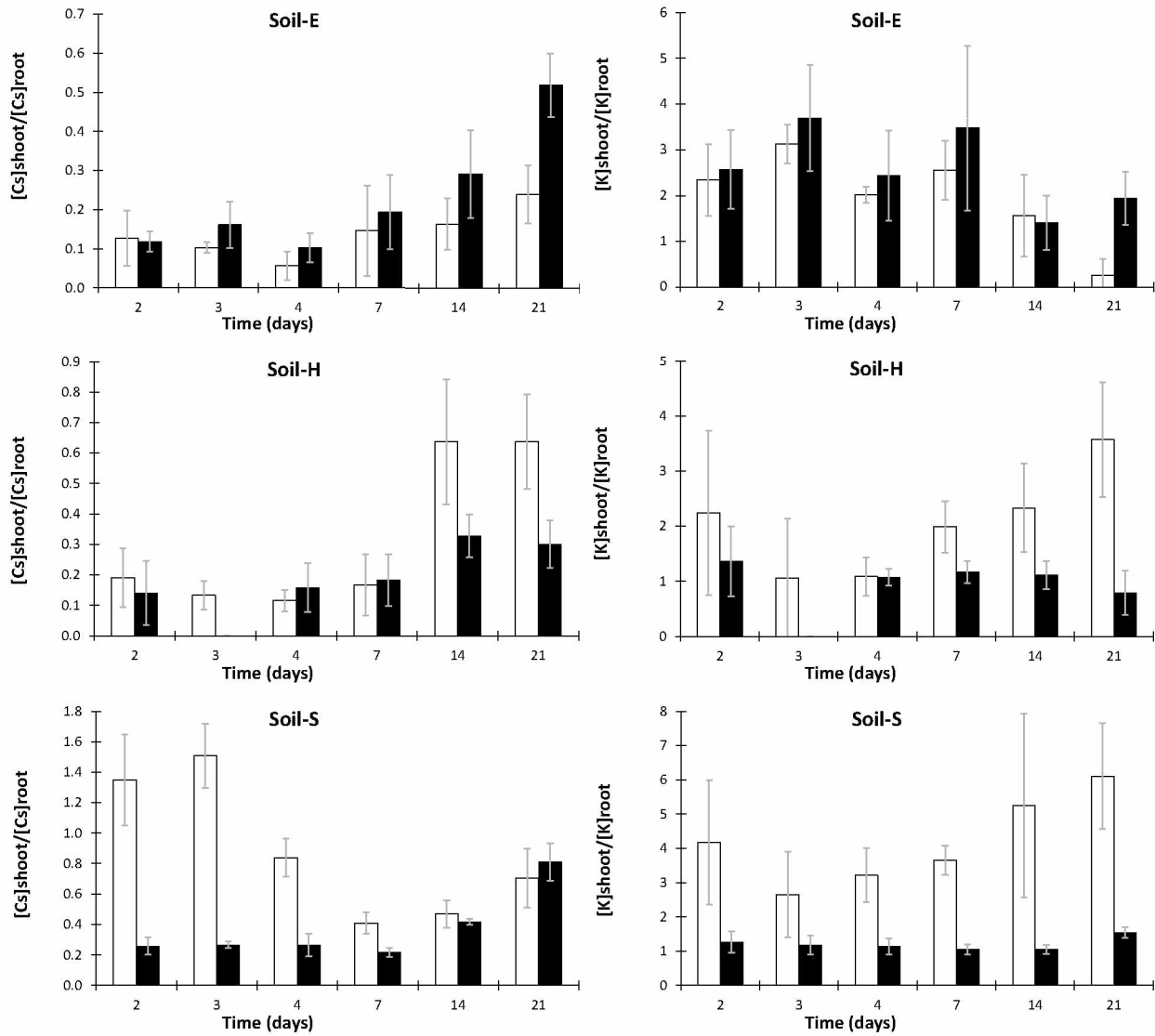
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851 *Figure S5: Concentration of potassium in the plant and its distribution (white: potassium concentration in*
 852 *the root; black: potassium concentration in the shoot; hatched: potassium concentration in the*
 853 *plant)*

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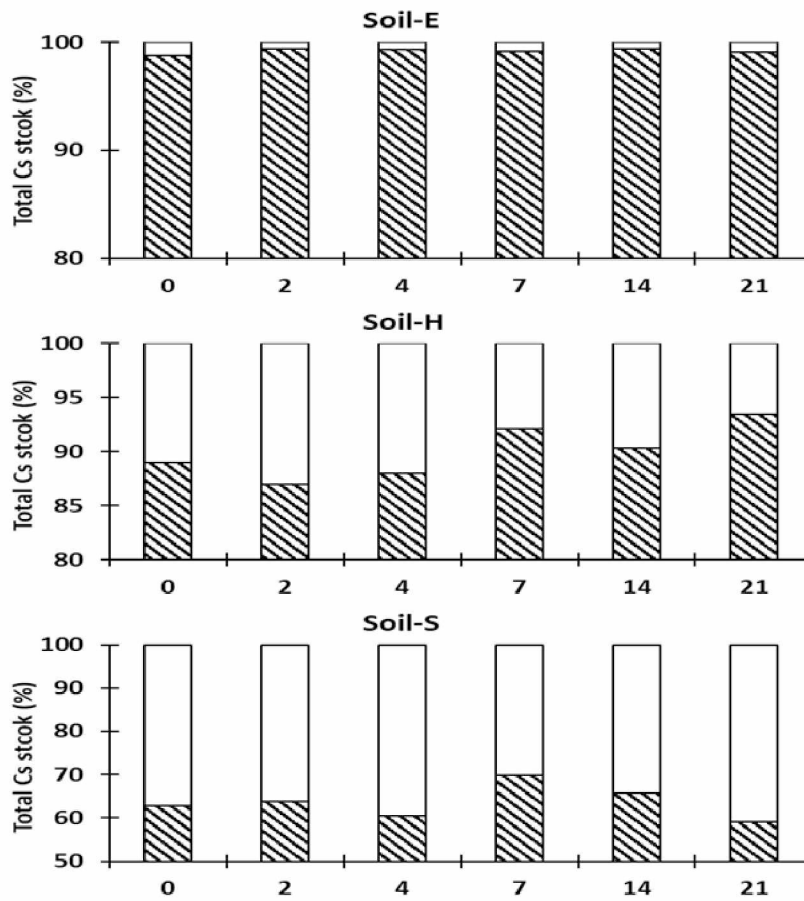


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857 *Figure S6: Concentration ratio in the shoot versus the root for cesium (left) and potassium (right) (white:*
 858 *millet; black: mustard)*

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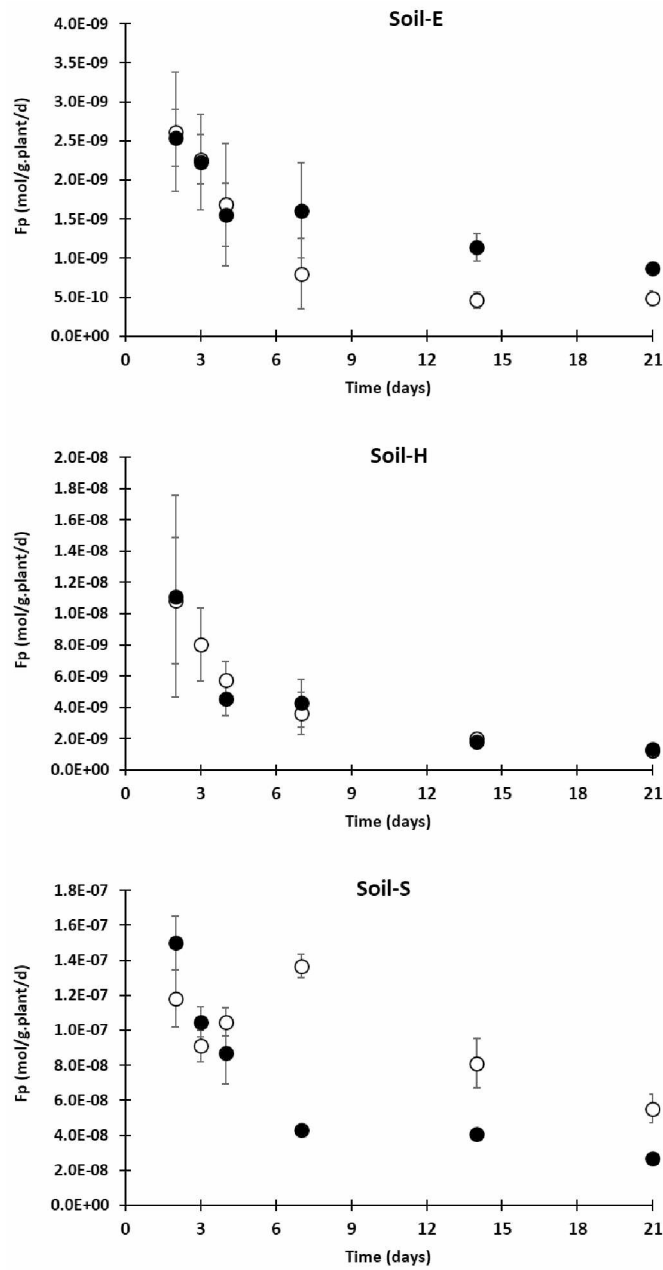
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862 *Figure S7: Variation in cesium quantities, expressed as a percentage of the initial cesium added to the*
863 *soil, in the available fraction (white) and in the fixed fraction (hatched) of the soils during the experiment*
864 *in unplanted Rhizotests*

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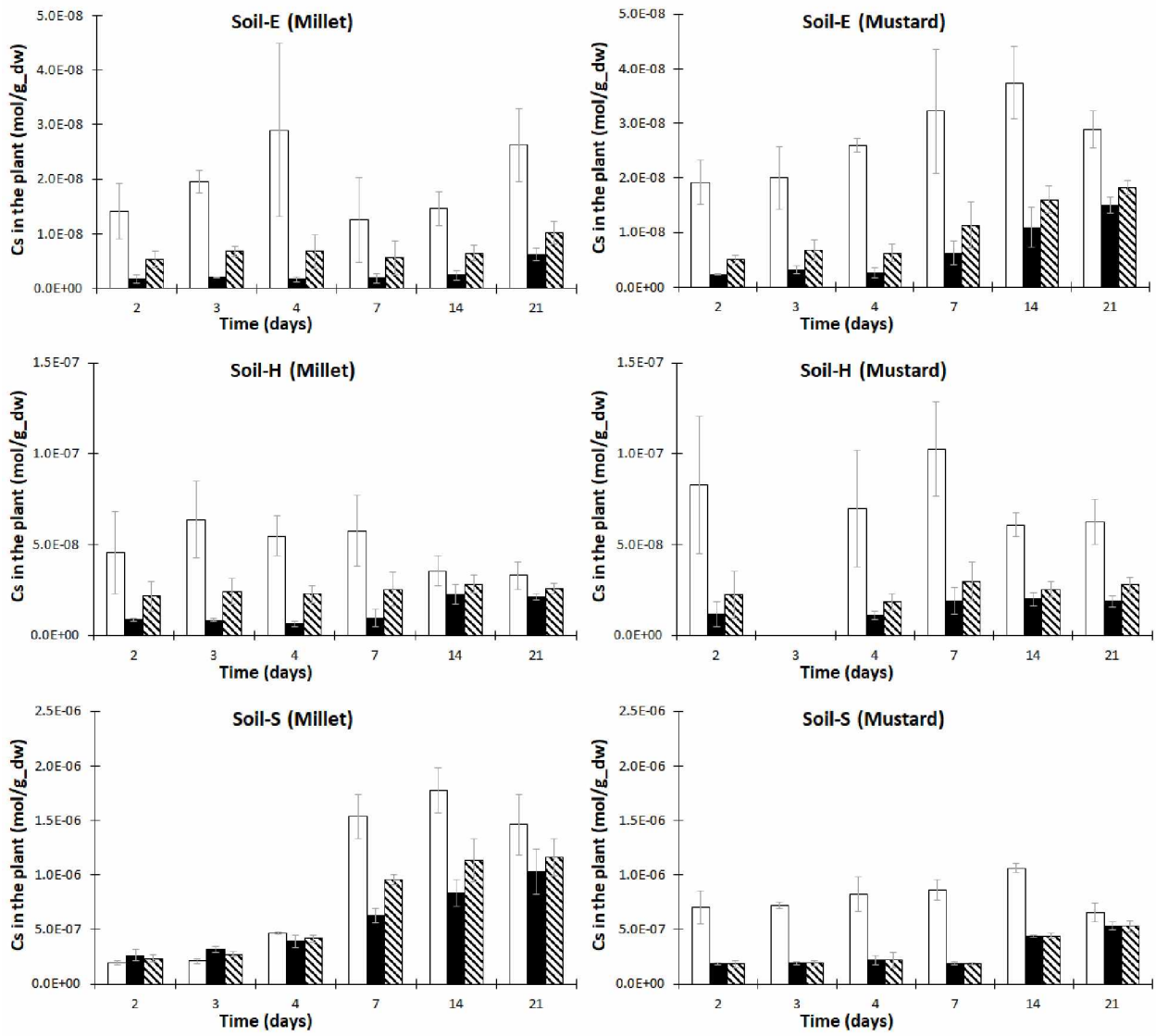
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868 *Figure S8: Cesium plant uptake rate for each experiment. Empty dots represent the millet experiment and*
 869 *filled dots represent the mustard experiment*

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874 *Figure S9: Concentration of cesium in the plant (white: cesium concentration in the root; black: cesium*
875 *concentration in the shoot; hatched: cesium concentration in the whole plant)*

876