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**Phosphatase-mediated bioprecipitation of lead by soil fungi**

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Running title: Phosphatase-mediated lead bioprecipitation by fungi
Summary

Geoactive soil fungi were examined for their ability to release inorganic phosphate (P$_i$) and mediate lead bioprecipitation during growth on organic phosphate substrates. Aspergillus niger and Paecilomyces javanicus grew in 5 mM Pb(NO$_3$)$_2$–containing media amended with glycerol 2-phosphate (G2P) or phytic acid (PyA) as sole P sources, and liberated P$_i$ into the medium. This resulted in almost complete removal of Pb from solution and extensive precipitation of lead-containing minerals around the biomass, confirming the importance of the mycelium as a reactive network for biomineralization. The minerals were identified as pyromorphite (Pb$_5$(PO$_4$)$_3$Cl), only produced by P. javanicus, and lead oxalate (PbC$_2$O$_4$), produced by A. niger and P. javanicus. Geochemical modelling of lead and lead mineral speciation as a function of pH and oxalate closely correlated with experimental conditions and data. Two main lead biomineralization mechanisms were therefore distinguished: pyromorphite formation depending on organic phosphate hydrolysis and lead oxalate formation depending on oxalate excretion. This also indicated species specificity in biomineralization depending on nutrition and physiology. Our findings provide further understanding of lead geomycology and organic phosphates as a biomineralization substrate, and are also relevant to metal immobilization biotechnologies for bioremediation, metal and P biorecovery, and utilization of waste organic phosphates.

Keywords: lead, pyromorphite, lead oxalate, fungi, phosphatase, geochemical modelling, biomineralization
Lead is one of the most widely found toxic metal pollutants in the environment, and arises from natural weathering of rocks and industrial activities, including mining and smelting of lead ores, manufacture of lead-acid batteries, ammunition waste and colouring elements in pigments (Flora et al., 2012). In recent years, the application of metal and mineral transformation capabilities of microbes in bioremedial treatments for metal contamination has been widely accepted (Ruby et al., 1994; Cotter-Howells, 1996; Sayer et al., 1999; 2001; Fomina et al., 2007, 2008; Debela et al., 2010; Rhee et al., 2012; 2014a,b; Wei et al., 2013; Gupta and Lu, 2013; Li et al., 2014; Gadd et al., 2014). Pyromorphite ($\text{Pb}_5(\text{PO}_4)_3X$ (X = OH, Cl, or F)) is one of the most stable lead minerals and its formation can reduce the bioavailability of lead in the environment (Cotter-Howells and Giddens, 1990; Ruby et al., 1994; Debela et al., 2010; Rhee et al., 2012; 2014a,b). It is a secondary lead mineral and can be formed by the weathering of galena (PbS) under oxidizing conditions although its formation may be limited by the availability of phosphorus (Cotter-Howells and Giddens, 1990). A commonly proposed strategy to immobilize Pb in the environment is by formation of pyromorphite by addition of an inorganic phosphate (P$_i$) source such as Ca-phosphate, orthophosphate, or polyphosphate (Cotter-Howells, 1996; Cotter-Howells and Caporn, 1996; Hashimoto et al., 2009; Dick et al., 2011). The elevation of P$_i$ concentration and combination with Pb$^{2+}$ and Cl$^-$ results in precipitation as pyromorphite (Ruby et al., 1994; Ma et al., 1995; Cotter-Howells, 1996; Cotter-Howells and Caporn, 1996). Under general geochemical conditions, the pyromorphite family $\text{Pb}_5(\text{PO}_4)_3X$, where X = F, Cl, Br, OH, are the most stable environmental Pb compounds ($\text{Pb}_5(\text{PO}_4)_3\text{F}$, $K_{sp} = 10^{-71.6}$, $\text{Pb}_5(\text{PO}_4)_3\text{Cl}$, $K_{sp} = 10^{-84.4}$,
Pb\(_5\)(PO\(_4\))\(_3\)Br, \(K_{sp} = 10^{-78.1}\), and Pb\(_5\)(PO\(_4\))\(_3\)OH, \(K_{sp} = 10^{-76.8}\) over a wide range of conditions (Chen et al., 2007; Chrysochoou et al., 2007). However, it has been clearly demonstrated that certain microbial metabolites, e.g. mycogenic organic acids, can affect pyromorphite stability (Sayer et al., 1999; Fomina et al., 2004; Manecki and Maurice, 2008; Debela et al., 2010; Topolska et al., 2014). The formation of lead oxalate from pyromorphite by several free-living and mycorrhizal fungi has been demonstrated (Sayer et al., 1999; Fomina et al., 2004).

Phosphorus is abundant in the environment, but most is in an insoluble form, and soluble \(P_i\) readily reacts with a variety of divalent and trivalent cations in soil or aquatic systems to form inorganic compounds or large insoluble complexes (Van Ho et al., 2002). Insoluble inorganic and organic-P compounds can be utilized by bacteria and fungi by, e.g. organic acid production and phosphatase enzyme activity respectively (Whitelaw, 2000; Chen et al., 2007). Microbes can import phosphate from phosphate-containing organic compounds by first cleaving C-O-P ester bonds with phosphatases that release \(P_i\) (Hyšek and Sărapatka, 1997; Hayes et al., 2000; Yadav and Tarafdar, 2001; Bull et al., 2002). Phosphate-solubilizing bacteria and fungi can solubilize inorganic phosphate-containing minerals, e.g. apatites and other phosphates (Sayer et al., 1995, 1997, 1999; Di Simine et al., 1998; Sayer and Gadd, 1997, 2001; Whitelaw, 2000; Jacobs et al., 2002a,b; Fomina et al., 2004, 2005; Ahemad et al., 2009; Topolska et al., 2014) but can also release phosphate from organic phosphate sources through phosphatase activity (Aickin et al., 1979; Macaskie et al., 1992, 1994, 2000; Yong and Macaskie, 1995; Boswell et al., 1999, 2001; Dick et al., 2011). Released phosphate from organic sources can precipitate with metals forming insoluble metal phosphates, a phenomenon first characterized in bacteria (Aickin et al., 1979; Macaskie et al., 1992, 1994, 2000; Yong and Macaskie,
1995; Boswell et al., 1999, 2001). Formation of metal phosphates has been observed in fungi, e.g. *Penicillium ochro-chloron* precipitated cupric oxalate and cupric phosphate (Crusberg, 2004), while *Penicillium cyclopium* removed up to 95% supplied lead in the presence of phosphate (Tsekoval et al., 2006). Rhee et al. (2012, 2014a,b) have shown fungal mediation of pyromorphite (Pbs(PO$_4$)$_3$Cl) formation from lead metal. Under conditions of Cu and Cd toxicity, the ectomycorrhizal fungus *Xerocomus chrysenteron* was exuded soluble protein and acid phosphatase that appeared to be involved in metal tolerance and precipitation (Zheng et al., 2009). *Laccaria laccata* and *Suillus bovinus* have also been shown to produce more acid phosphatase in the presence of high concentrations of Cr (VI) (Raman et al., 2002).

The objective of this work was to determine the possible role of fungal phosphatases in the bioprecipitation of lead phosphate when organic phosphate substrates are the only substrate source of phosphorus. The pH and P dependence of lead phosphate formation, and the significance of excreted oxalate in lead biomineralization was also investigated, coupled with geochemical modelling of lead and lead mineral speciation as a function of pH and oxalate.
Results

Growth and $\text{P}_\text{i}$ release from different organic-P sources

All fungi were able to grow in organic phosphorus-amended MCD media at both pH 5 and pH 8. Negligible growth was observed in MCD medium without any phosphorus source. With equivalent amounts of P in the MCD medium, all fungi grew better and released more $\text{P}_\text{i}$ into the medium when amended with 30 mM G2P than with 5 mM PyA. Among the test fungi, *A. niger* grew well with G2P and released up to 23 mM $\text{P}_\text{i}$ from 30 mM G2P after 30 days incubation at 25°C. There were no significant differences in the amount of $\text{P}_\text{i}$ released by *A. niger* and *P. javanicus* when grown with G2P at the different initial pH values. *P. javanicus* had a higher ability to hydrolyze PyA than the other test species, with 8.5 mM $\text{P}_\text{i}$ released from 5 mM PyA after growth in MCD medium for 30 days at both initial pH values (Fig. 1). There were no significant differences in growth rate and $\text{P}_\text{i}$ release by the organisms when grown at different initial pH values except for *B. caledonica* and *R. rubescens*. The latter released more $\text{P}_\text{i}$ from the organic phosphorus sources in acidic rather than in alkaline conditions. *B. caledonica* and *R. rubescens* released up to 18 and 13 mM $\text{P}_\text{i}$, respectively, from 30 mM G2P in acidic conditions, compared with 11 and 7 mM $\text{P}_\text{i}$, respectively, in alkaline conditions. From analysis of the fungal growth rates and $\text{P}_\text{i}$ release from organic-P, *A. niger* and *P. javanicus* were selected for detailed experiments.

Growth and $\text{P}_\text{i}$ release from different organic-P sources in the presence of Pb by *A. niger* and *P. javanicus*
In the presence of Pb(NO$_3$)$_2$, growth of $A.$ niger was reduced, while $P.$ javanicus showed a relatively better tolerance in lead-amended MCD medium (Table 1). Tolerance indices (TI) were used to compare the biomass yields of $A.$ niger and $P.$ javanicus grown on MCD medium with or without 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA (Table 1). A TI value lower than 100% indicates growth inhibition. Biomass yields of $A.$ niger were markedly reduced in the presence of 5 mM Pb(NO$_3$)$_2$ when grown with 30 mM G2P, with growth inhibition resulting on 5 mM Pb(NO$_3$)$_2$ and 5 mM PyA. For $P.$ javanicus, there was some inhibition of biomass yield over the first 10 days incubation, but after this time, biomass yields were generally larger than on control medium. The pH of all MCD media was adjusted to pH 6 before inoculation. The pH value of medium containing 30 mM G2P dropped to pH 3.1 after growth of $A.$ niger for 30 days while the corresponding pH for $P.$ javanicus in the same culture medium increased to pH 6.8. The pH values of medium containing 5 mM PyA showed similar trends after growth of $A.$ niger and $P.$ javanicus (Table 1). P$_i$ concentrations in MCD medium containing 30 mM G2P or 5 mM PyA increased with time during growth of $A.$ niger and $P.$ javanicus. $A.$ niger released around 25-26 mM P$_i$ from 30 mM G2P and 5 mM PyA respectively after 30 days growth (Table 1). Similar concentrations of free P$_i$ were released by $P.$ javanicus. However, $P.$ javanicus released more P$_i$ at an earlier stage of growth than $A.$ niger, with around 9-10 mM P$_i$ being released from $A.$ niger and around 15 mM P$_i$ being released from $P.$ javanicus after 20 days (Table 1).

**Pb removal from solution and the formation of lead mineral phases**
Both *A. niger* and *P. javanicus* were able to remove almost all of the lead from media amended with 5 mM Pb(NO$_3$)$_2$ and the organic-P sources (Table 1). Both *A. niger* and *P. javanicus* removed most of the Pb$^{2+}$ from solution after 10 days and showed increasing Pb accumulation with time (Table 1).

Both *A. niger* and *P. javanicus* were able to grow in the presence of 5 mM Pb(NO$_3$)$_2$ and precipitated secondary minerals (Fig. 2). In contrast to *A. niger* from control Pb-free MCD medium (Fig. 2A), extensive secondary mineral formation occurred when *A. niger* was grown with 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P (Fig. 2B). Some secondary minerals with a similar shape were also found on the biomass of *A. niger* grown with 5 mM Pb(NO$_3$)$_2$ and 5 mM PyA (Fig. 2E). For *P. javanicus*, the produced secondary minerals showed a completely different shape from those associated with *A. niger*, with arborescent and blade-like shapes above the hyphae (Fig. 2C,F) with some arborescent and globular (Inset Fig. 2C).

**Energy-dispersive X-ray analysis (EDXA)**

EDXA revealed the elemental composition of the secondary minerals found on the fungal hyphae. The minerals associated with *A. niger* after growth with Pb(NO$_3$)$_2$ and G2P showed peaks for carbon, oxygen, potassium and lead as the main elements detected (Fig. 3A). The minerals produced by *P. javanicus* grown with Pb(NO$_3$)$_2$ and G2P (Fig. 3B) showed peaks for carbon, oxygen, sodium, phosphorus and lead as the main elements. The minerals produced by *A. niger* grown with Pb(NO$_3$)$_2$ and PyA that showed a cylindrical form showed carbon, oxygen, sodium, potassium and lead (Fig. 3C). The minerals produced by *P. javanicus* grown with Pb(NO$_3$)$_2$ and PyA (Fig.
3D) showed peaks for carbon, oxygen, sodium, aluminium, phosphorus and lead as the main elements present.

Transmission electron microscopy (TEM) revealed the presence of electron-dense deposits around the biomass of both *A. niger* and *P. javanicus* after growth with 5 mM Pb(NO₃)₂ and 30 mM G2P (Fig. 4). Compared to the electron-dense granules found with *P. javanicus*, the amount of crystals found with *A. niger* seemed of lower abundance. These observations are consistent with the presence of the secondary minerals found with SEM.

**X-ray powder diffraction (XRPD)**

The secondary mineral precipitates associated with *A. niger* and *P. javanicus* grown with lead and G2P showed the presence of well-crystallized compounds with an excellent match to reference patterns for lead oxalate (PbC₂O₄) and pyromorphite (Pb₅(PO₄)₃Cl) (Fig. 5). Lead oxalate was detected from both *A. niger* and *P. javanicus*, but pyromorphite was only detected with *P. javanicus* (Fig. 5).

**Geochemical modelling**

The predominance diagrams for chemical species of lead can be simulated by PHREEQC, and this software was used to generate a diagram for the bioprecipitation of lead oxalate and pyromorphite. For lead, the dominant minerals found in the simulated systems were simple oxides, carbonates and phosphates. Lead minerals differ widely in water solubility depending on the presence of H⁺, HCO₃⁻, PO₄³⁻, SO₄²⁻, and the total ionic charge of the solution. The diagram shows
the most dominant mineral species of lead theoretically present in the MCD medium (Fig. 6). The data was calculated for a temperature of 25°C and with total dissolved lead in the presence of dissolved chloride, carbonate, phosphate, nitrate and sulfate. Fig. 6A indicates that pyromorphite dominates over the range pH 2-11 in the absence of oxalic acid, Pb(H₂PO₄)₂ dominates at pH values < 2, Pb(OH)₂ dominates over pH 11-13, and Pb(OH)₄²⁻ occurs at pH values > 13. In the presence of oxalic acid, the situation is different and lead oxalate and lead hydroxide become the two main species in the system (Fig. 6B). Lead oxalate dominates over the pH-range 2-7, while lead hydroxide dominates over pH 7-13. Pb²⁺ and Pb(OH)₄²⁻ dominate at pH< 2 and pH >13, respectively. In Fig. 6C, the mineral stability diagrams as a function of pH and oxalate activity show the dominance of lead oxalate at a pH < 5 and at an oxalate activity >0.1 mM. These pH-ranges correspond with the pH values measured in the experimental conditions (Table 1). In contrast, anglesite can become dominant over the same acidic pH range when the oxalate concentration is lower than 0.1 mM. Pyromorphite is dominant over the approximate range of pH 4.8-8 even at high concentrations of oxalate. At pH > 8, lead hydroxide becomes dominant at any oxalate concentration. The presence of pyromorphite in the P. javanicus-organic-P-Pb system simulation and the presence of lead oxalate in the A. niger-organic-P-Pb system closely correlates with the XRPD data, confirming that the theoretical simulation closely agrees with experimental findings. It is important to note that with the function of pH and Eₚₒ, the concentration of lead in the aqueous system strongly affects the formation of the different lead species, while aqueous Pb complexes can also undergo several protonation or deprotonation reactions together with polymerization to form various aqueous species of lead.
The availability of P is one of the most crucial determinants of microbial growth (Eide, 1998; Whitelaw, 2000). However, a large proportion of the phosphorus pool in nature is insoluble and this raises the question of how organisms obtain P from organic and insoluble phosphate sources (Wenzel et al., 1994; Whitelaw, 2000; Fomina et al., 2004). The dissolution of inorganic phosphates by fungi is well known and this can result in release of the associated metals (Lang and Kaupenjohann, 2003; Fomina et al., 2004; Hashimoto et al., 2009; Gadd, 2007, 2010). The possibility of utilizing organic phosphate sources in the environment holds some promise for toxic metal bioremediation or biorecovery through metal phosphate precipitation, and a large number of microorganisms, including bacteria and fungi, can produce various extracellular phosphatases (Schurr and Yagi, 1971; Sano et al., 1999; Millán, 2006). Fungal phosphatases are able to hydrolyse different organic-P sources releasing inorganic phosphate (P\textsubscript{i}) (Field and Schekman, 1980; Dick et al., 2011). The enzymes can be classed as alkaline phosphatases with pH optima above 7, and acid phosphatases with pH optima below 7. Acid phosphatases can cleave the C-O-P ester bonds, which do not penetrate the plasma membrane, and release P\textsubscript{i} (Yadav and Tarafdar, 2001; Hayes et al., 2000). These enzymes are therefore usually associated with the cell walls of fungi (Field and Schekman, 1980; Novick et al., 1980; González et al., 1993; Ito et al., 2007). Furthermore, phytase enzymes, which are meso-inositol hexaphosphate phosphohydrolases, catalyse the step-wise phosphate splitting of phytic acid (inositol phosphate: IP\textsubscript{6}) or phytate to lower inositol phosphate esters (IP\textsubscript{5}-IP\textsubscript{1}) and P\textsubscript{i} (Hayes et al., 2000). Aspergillus and Penicillium spp. have been shown to produce extracellular acid phosphatases that hydrolysed
extracellular organic phosphates in the presence of copper which enhanced the removal of copper from solution (Haas et al., 1991; Tsekova et al., 2000; 2002). The Pi released can be accumulated within cells via specific transport systems, but can also react with and precipitate toxic metals as insoluble metal phosphates (Crusberg, et al., 2003).

In this work, organic phosphates were supplied as the sole P source. After growth in MCD medium amended with organic phosphorus and Pb(NO₃)₂, both A. niger and P. javanicus removed nearly all the soluble Pb²⁺ in the medium. This appeared to be mainly by two different mechanisms, (i) phosphatase-mediated hydrolysis of the phosphate source leading to phosphate precipitation and (ii) oxalate excretion leading to oxalate formation. The precipitation of metal oxalates has previously been considered as a means of metal detoxification in fungi (Manley and Evans, 1986; Müller et al., 1995; Banfield, 1999; Gadd, 1999; 2004; Fomina et al., 2004; Wei et al., 2013; Gadd, et al., 2014). Previous research has demonstrated the effect of mycogenic organic acids on the solubility of pyromorphite and other minerals (Sayer et al., 1999; Fomina et al., 2004). Pyromorphite can be dissolved by organic acid-producing fungi, such as A. niger, and this can release free Pi and Pb from pyromorphite when it is the sole source of P (Sayer et al. 1999). In contrast, other fungi are capable of mediating pyromorphite formation from sources of metallic lead (Rhee et al., 2012; 2014a,b). In this work, formation of pyromorphite only occurred with P. javanicus grown with organic-P in the presence of lead while lead oxalate was found with both A. niger and P. javanicus.

Oxalate can be secreted by many fungi and this can mediate efficient ligand-promoted Pb mobilization and immobilization (Sayer et al., 1999; Fomina et al., 2004). The dominance of lead oxalate rather than pyromorphite is the result of ion
competition with the soluble P (either from the organic-P or the P<sub>i</sub> released from organic-P), biosorption to fungal biomass, the pH of the system and Pb complexation with dissolved organic carbon or organic acids (Lang and Kaupenjohann, 2003; Gadd, 2004; Ryan <em>et al.</em>, 2004; Hashimoto <em>et al.</em>, 2009). When additional phosphate was added to Pb-contaminated soil without any microorganisms present, about 70% of the Pb species present were not transformed into pyromorphite or indeed any lead phosphate (Hashimoto <em>et al.</em>, 2009), which may emphasize the importance of fungal and other microbial activities in the formation of pyromorphite and other lead secondary minerals (Rhee <em>et al.</em>, 2012; 2014a,b).

Previous research has shown that pyromorphite formation in soil can be inhibited by the presence of dissolved organic carbon (Lang and Kaupenjohann, 2003) and organic matter (Hashimoto <em>et al.</em>, 2009) through organic-metal complex formation. Commonly occurring low molecular weight organic acids (LMWOA) secreted by fungi in the rhizosphere were examined for dissolution of pyromorphite at a 100 µM concentration. The means of the highest amounts of Pb (µM Pb) in solution followed the order: oxalic acid (17.6) > citric acid (6.2) > malic acid (5.6) > acetic acid (3.0) (Debela <em>et al.</em>, 2010). Solution pH, concentration of the carbon, phosphorus and nitrogen sources, the buffering capacity of the medium, the presence or absence of certain metals and some trace elements can influence organic acid secretion (Sayer and Gadd, 1997, 2001; Gharieb and Gadd, 1999; Gadd, 1999; Gadd <em>et al.</em>, 2012; 2014). Solution pH strongly influences the activities of metals and the de/protonation of LMWOA (Debela <em>et al.</em>, 2010). However, solution pH alone cannot explain the lead secondary mineral formation observed in this study but may be a contributing factor in the formation of lead oxalate. The high affinity of oxalate for Pb seems to be a major factor in lead oxalate formation. Oxalic acid excretion can be strongly
influenced by the presence of toxic metals (Fomina et al., 2004). For instance, during growth of *A. niger* in the presence of Co\(_3\)(PO\(_4\))\(_2\) and Zn\(_3\)(PO\(_4\))\(_2\), there was increased production of gluconic acid and citric acid respectively (Sayer and Gadd, 2001). Generally, fungi acidify the medium during growth, but such acidification can depend on the nitrogen source, tending to result with NH\(_4^+\) as nitrogen source rather than NO\(_3^-\) (Lapeyrie, 1988; Wenzel et al., 1994; Whitelaw, 2000; Gharieb and Gadd, 1999; Fomina et al., 2004). The mechanisms for lead secondary mineral formation therefore appear to include: (1) organic acid secretion by fungi decreasing the solution pH and forming surface lead complexes through adsorption to fungal surfaces, (2) the saturation state of the solution being altered by the generation of organic acids with respect to mineral formation, (3) metal speciation being greatly influenced by the presence of organic acids and Pi released by the fungal phosphatases, and (4) pH control of metal hydrolysis and complexation (McBride, 1994). Fungal cell walls have an important role in metal bioprecipitation and biotransformation providing many binding sites for toxic metals (Van Tichelen, et al., 2001; Adriaensen et al., 2004; Fomina et al., 2004; Gadd, 1993, 2007, 2009; Gadd et al., 2014; Li et al., 2014). The range in Pb:P molar ratios suggests that the formation of pyromorphite was stoichiometric.

Lead oxalate is stable and dominant in the medium as long as the activity of oxalate remains high (>0.1 mM) and the pH remains low (pH<5), which are the conditions close to fungal growth in our experiments. Lead oxalate tends to be stable in acidic environments with a low sulfur concentration which avoids the formation of anglesite (PbSO\(_4\)). Anglesite, and pyromorphite formation, has been shown to occur with several yeast strains incubated in Pb\(^{2+}\) - and SO\(_4^{2-}\)-containing media (Liang et al. 2015a). As oxalate activity decreases or the medium become more alkaline, other
minerals can become more stable. Because sulfate concentration is generally higher in soils than oxalate, lead oxalate in soil is not so stable. Over a wide pH range, there is a possibility that lead oxalate could be transformed into pyromorphite in the presence of phosphate. The conversion of lead oxalate to pyromorphite suggests that a dynamic equilibrium exists between chemical and biological processes and even compared to lead oxalate ($\log K_{sp} = -8.07$), chloropyromorphite ($\text{Pb}_5(\text{PO}_4)_3\text{Cl}$) ($\log K_{sp} = -84.4$) and hydroxy pyromorphite ($\text{Pb}_5(\text{PO}_4)_3(\text{OH})$) ($\log K_{sp} = -76.8$) are far less soluble than many other lead minerals (Shevade et al., 2001; Scheckel and Ryan, 2002; Fomina et al., 2004).

In conclusion, this work has shown that filamentous fungi are capable of phosphatase-mediated bioprecipitation of lead when utilizing an organic-P-containing substrate. This complements first discoveries of this phenomenon in bacteria (Aickin et al. 1979) and yeasts (Liang et al. 2015a). Fungal biomineralization was, however, species specific and therefore must depend on the physiology of the biomineralizing organism, and the changes it can effect in its environment. Pyromorphite was only produced by $P. javanicus$, this organism previously shown to be capable of pyromorphite formation from lead metal (Rhee et al. 2012, 2014a,b) while only lead oxalate was produced by $A. niger$, a result of significant oxalate secretion properties of this organism (Gadd, 1999). Lead speciation modelling closely correlated with experimental results and the physico-chemical conditions of the media. It is likely therefore, that fungal species specificity of biomineralization depends on nutritional characteristics and metabolism, such as the substrate and the nature and amount of organic acids excreted, the inorganic nitrogen source, as well as phosphatase activity in the case of organic-P sources. Filamentous fungi have received lower attention than bacteria regarding phosphate precipitation but it has been shown that
fungi can precipitate U-containing phosphate minerals during growth in the presence or inorganic or organic sources of P (Fomina et al., 2007, 2008; Liang et al., 2015b).

The ability of fungi to precipitate metal phosphates from organic P resources provides further understanding of fungal interactions with toxic metals and minerals in polluted sites and their roles in the biogeochemical cycling of metals as well as P. Metal bioprecipitation is also relevant to new approaches in bioremediation of polluted environments, as well as metal biorecovery and bioprocessing, and the manufacture and use of new biomaterials.
Experimental procedures

Organisms and media

Aspergillus niger van Tieghem (ATCC 201373), Beauveria caledonica Bisset and Widden 4 (originally supplied by D. Genney (CEH Merlewood collection)), Rhizopogon rubescens Tulasne (kindly provided by Dr. H. Wallander) and Paecilomyces javanicus (Friedrichs & Bally) A.H.S. Brown & G. Smith were used. These fungi have significant abilities in mineral and toxic metal biotransformations (Sayer et al., 1999; Fomina et al., 2007; 2008; Rhee et al., 2012; 2014a,b; Wei et al., 2013; Gadd et al., 2014). Test fungi were grown in modified Czapek-Dox liquid medium (MCD) amended with 30 mM glycerol 2-phosphate disodium salt hydrate (C₃H₇Na₂O₆P·xH₂O) (G2P) (Sigma-Aldrich, St. Louis, MO, USA) or 5 mM phytic acid sodium salt hydrate (C₆H₁₈O₂₄P₆·xNa⁺·yH₂O) (PyA) (Sigma-Aldrich, St. Louis, MO, USA) as sole phosphorus sources in 250-ml Erlenmeyer conical flasks containing 100 ml nutrient medium on an orbital shaking incubator (Infors Multitron Standard, Rittergasse, Switzerland) at 125 rpm at 25°C in the dark. 10 x 5 mm diameter inoculum plugs, cut from the margins of actively growing fungal colonies using a sterile cork borer (autoclaved at 121°C for 15 min) were used as inoculum. A. niger was grown for 4 d, B. caledonica was grown for 5 d, while P. javanicus and R. rubescens were grown for 10 d prior to experimental subculture. MCD medium consists of (L⁻¹ Milli-Q water (Merck Millipore, Billerica, Massachusetts, USA)): D-glucose 30 g (Merck, Readington Township, NJ, USA), NaNO₃ 3 g (Sigma-Aldrich), MgSO₄·7H₂O 0.5 g (VWR, Radnor, PA, USA), KCl 0.5 g (Sigma-Aldrich), Fe(NO₃)₃·9H₂O 0.01 g (Sigma-Aldrich). G2P and PyA were dissolved
separately in Milli-Q water and sterilized by membrane filtration (cellulose nitrate, 0.2 µm pore diameter, Whatman, Maidstone, Kent, UK) and added to autoclaved MCD medium (121°C, 15 min) at room temperature to give final concentrations of 30 mM G2P and 5 mM PyA. After autoclaving, the pH of liquid medium was adjusted to pH 5 using sterile 1 M HCl or to pH 8 using sterile 1 M NaOH.

*A. niger* and *P. javanicus* were chosen as test organisms for detailed experiments. These were grown in MCD amended with 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA as sole phosphorus sources in 250-ml conical flasks containing 100 ml nutrient medium on an orbital shaking incubator at 125 rpm at 25°C in the dark. Pb(NO$_3$)$_2$, G2P and PyA were dissolved separately in Milli-Q water and sterilized by membrane filtration (cellulose nitrate, 0.2 µm pore diameter, Whatman, Maidstone, Kent, UK) and added to autoclaved MCD medium (121°C, 15 min) at room temperature, to give 5 mM Pb(NO$_3$)$_2$, 30 mM G2P and 5 mM PyA final concentrations. All experiments were conducted at least in triplicate.

**Tolerance indices (TI), P$_i$ release, lead measurement and pH analysis**

Test fungi were grown in MCD with or without 30 mM G2P or 5 mM PyA at pH 5 and pH 8. 10 mL aliquots of supernatant were collected at time intervals for up to 30d for analysis of P$_i$ and pH measurements.

Inorganic P$_i$ released during growth was quantified spectrophotometrically after reaction in the malachite green assay (Irving and McLaughlin, 1990). For these phosphate measurements, 15 µL aliquots of supernatant were sterilized by membrane filtration (cellulose nitrate, 0.2 µm pore diameter, Whatman, Maidstone,
Kent, UK), and added to wells in a 96-well plate with 185 µL Milli-Q water followed by addition of 100 µL malachite green reagent. After mixing for 15 min, the absorbance at 620 nm was read using a RosysAnthos 2001 microplate reader, and calculations were carried out using the manufacturer’s Windows-based control and evaluation software (AnthosLabtec Instruments, Wals-Siezenheim, Austria). For the malachite green background comparison, 200 µL Milli-Q water was mixed with 100 µL malachite green reagent standard in individual wells in the 96-well plate giving a final volume of 300 µL. After incubating the plate for 15 min, the absorbance at 620 nm was determined as described previously. The pH of supernatants was measured using a pH 210 Microprocessor pH Meter (Hanna Instruments, Woonsocket, RI, USA). All experiments were conducted at least in triplicate.

For examining the role of fungal phosphatase in lead bioprecipitation, test fungi were grown in MCD with 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA in 250-ml conical flasks containing 100 ml nutrient medium on an orbital shaking incubator at 125 rpm at 25°C in the dark. Fungal biomass was harvested by centrifugation at 4000 rpm (4880g) for 30 min and supernatants were obtained by filtering through membrane filters (0.2 µm pore diameter, Whatman, Maidstone, Kent, UK) at appropriate time intervals and analysed for lead concentration, and pH. Metal tolerance was evaluated using a tolerance index (TI) as follows: (dry weight of lead-exposed mycelium/dry weight of control mycelium x 100%) (Sayer et al., 1995; Wei et al., 2013). Fungal biomass was oven-dried at 105°C to constant weight and then ground to a powder using a pestle and mortar (Milton Brook, Dorset, UK). Samples for lead analysis were prepared by digestion of 50 mg dried biomass powder which was placed in a 50 mL glass test tube to which 3 mL concentrated nitric acid (15.8 M) was added with heating at 100°C until the solution was clear (~ 4 h). The resulting
digest was then diluted to a suitable concentration with Milli-Q water and passed
through a 0.2 µm syringe filter (Whatman, Maidstone, Kent, UK) prior to analysis.
Samples were stored at 4°C before analysis of metal content by atomic absorption
spectrophotometry (AAS, Perkin Elmer, AAnalyst 400) with reference to appropriate
standards. Calibration standards were prepared from 1000 mg L⁻¹ AAS standard
solutions for lead (Sigma-Aldrich Company Ltd., Gillingham, Dorset, UK). Any
difference in means between treatments was assessed using one way analysis of
variance (ANOVA) to a 0.05 significance level.

Examination of fungal biominerals produced in the presence of lead

Secondary mineral formation in association with fungal biomass grown with
Pb(NO₃)₂ and G2P or PyA was examined using scanning electron microscopy (SEM).
Fungal pellets grown for 30 d were cut in half using a sterile scalpel (Swann-Morton,
Sheffield, UK) and fixed in 2.5 % (v/v) triple-distilled glutaraldehyde in 5 mM 1,4-
piperazine N,N' bis (2-ethane sulphonic acid) (PIPES) buffer, pH 7.2, for at least 24 h
at room temperature. The pH of 5 mM PIPES was adjusted using 1 M NaOH using a
Corning pH meter 120 (Corning Incorporated, Corning, NY 14831, USA). After
fixation, samples were rinsed twice in 5 mM PIPES buffer, pH 7.2 (15 min per rinse)
and then dehydrated through a graded ethanol series (50-100 % (v/v), 15 min per
step). Samples were then critical point dried using a liquid CO₂ BAL-TEC CPD 0.30
critical point dryer (BAL-TEC company, Canonsburg, USA) and subsequently
mounted on aluminium stubs using carbon adhesive tape and stored in a desiccator
at room temperature. Prior to electron microscopy, samples were coated with 25nm
Au/Pd using a Cressington 208HR sputter coater (Ted Pella, Inc., Redding, CA, USA)
and examined using a Philips XL30 environmental scanning electron microscope (ESEM) (Philips XL 30 ESEM FEG) operating at an accelerating voltage of 15kV. Secondary minerals formed on the fungal hyphae were examined for elemental composition using energy-dispersive X-ray analysis (EDXA) before Au/Pd coating the samples in order to exclude the Au/Pd peak which overlaps P/Cl peaks. Spectra were acquired using a Phoenix EDXA analysis system embedded within the environmental scanning electron microscope (Philips XL30 ESEM FEG) operating at an accelerating voltage of 20kV. X-ray powder diffraction (XRPD) was used to identify the secondary minerals produced. Diffraction patterns were recorded from 3 to 60° 2-θ using Ni-filtered Cu K-alpha radiation, and scanning from 3-60° 2-theta counting for 300 seconds per step on a Panalytical X-pert Pro diffractometer using a X-celerator position sensitive detector. Mineral phases were identified by reference to patterns in the International Centre for Diffraction Data Powder Diffraction File (PDF).

For transmission electron microscopy (TEM), small cubes (~ 1 mm³) from fungal pellets grown in liquid media were cut using a clean scalpel. These were fixed in 2.5% (v/v) triple distilled glutaraldehyde in 5 mM PIPES buffer, pH 6.5, overnight at room temperature, and then washed four times (15 min/wash) with pH 6.5, 5 mM PIPES buffer. Dehydration was then performed through a 25-100% (v/v) ascending series of ethanol in sterile distilled water, samples being left overnight at each stage. L. R. Whyte resin (Agar Scientific Ltd, Stansted, UK) in absolute ethanol was used to infiltrate samples on a rotary mixer overnight after which time 100% resin was substituted twice, samples again being incubated overnight. After final infiltration, the small cubes containing fungal colonies were placed in gelatin capsules with fresh resin and polymerized at 60°C for 1 d. Ultrathin sections were cut on a Reichert
OMU-3 microtome (C. Reichert, Vienna, Austria) and mounted on formvar-coated copper grids. Unstained and stained sections (uranyl acetate and lead citrate) were examined on a Jeol-1200 EX transmission electron microscope (Jeol Ltd, Welwyn Garden City, UK).

**Modelling of lead speciation by PHREEQC**

Geochemical software (PHREEQC, Ver. 3 (Parkhurst and Appelo, 2013) was used to calculate the chemical equilibria of chemical and mineral species of lead, phosphorus and oxalate to produce a geochemical model for the biotransformation of pyromorphite and lead oxalate. The results are shown as predominance diagrams calculated by Phreeplot, the graphic supporting software for PHREEQC (Andres et al., 1993; 1994; Fowle et al., 2000; Kinniburgh and Cooper, 2004; 2014). The available Minteq v4 database was integrated with critically selected stability constants of metal-oxalate complexes taken from the database of the National Institute of Standards and Technology (NIST), Ver. 8 (Martell et al., 2004). There are some previous studies that characterized thermodynamic data for pyromorphite from which some data can be used for construction of the predominance diagrams for lead species under specific conditions (Sayer et al., 1999; Shevade et al., 2001). Total concentrations for Pb and P in the system were assumed to be 5 and 30 mM respectively, and 30 mM for oxalate, and the concentrations of other elements were taken to bethe same valuesas present in MCD medium. The oxalate value took into account previous measurements of oxalic acid excreted by A. niger, with 15.4mM being recorded after 10 d incubation at 25°C in Czapek-Dox liquid medium containing nitrate as a nitrogen source (Gharieb and Gadd, 1999).
Statistical analysis

All data presented in the paper are the means of at least three replicates and error bars represent one standard error either side of the mean. SigmaPlot, version 12.5, was used to perform statistical analyses. One-way ANOVA tests on means were performed for dry weight, the malachite green P$_i$ assay, pH and lead concentration measurements.

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culture filtrates of Aspergillus niger grown in the absence or presence of insoluble

compounds by soil fungi: development of a screening method for solubilizing ability

occurring metal-bearing minerals, limescale and lead phosphate by Aspergillus niger.


**Figure Legends**

**Fig. 1.** $P_i$ release from different P sources by *A. niger*, *B. caledonica*, *P. javanicus* and *R. rubescens* grown in MCD liquid media for 30 d at initial pH 5 and 8 at 25°C. The figure shows $P_i$ release in MCD medium without any added phosphorus source at pH 8 (■) and pH 5 (□), MCD medium amended with 30 mM G2P at pH 8 (□) and pH 5 (□) and MCD medium amended with 5 mM PyA at pH 8 (■) and pH 5 (□). All organisms were grown in the dark on an orbital shaking incubator at 125 rpm. Values shown are means of at least three measurements and the bars indicate typical standard deviations of about 5%.

**Fig. 2.** Scanning electron microscopy of biominerals formed by *A. niger* and *P. javanicus* grown in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA-amended MCD medium. (A) Control *A. niger* hyphae grown in Pb-free 30 mM G2P-amended MCD medium, scale bar = 50 µm. (B,C) Biominerals formed in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P-amended MCD medium after growth of (B) *A. niger* and (C) *P. javanicus* for 30 days, scale bars: B,C = 50 µm. Inset C is a higher magnification image of the area indicated by the square (scale bar = 5 µm). (D) Control *P. javanicus* hyphae grown in Pb-free 5 mM PyA-amended MCD medium for 30 days, scale bar = 20 µm. (E,F) Biominerals formed in 5 mM Pb(NO$_3$)$_2$ and 5 mM PyA-amended MCD medium after growth of (E) *A. niger* and (F) *P. javanicus* for 30 days, scale bars: E,F = 20 µm. All organisms were grown for 30 days at 25°C in the dark on an orbital shaking incubator at 125 rpm. Typical images are shown from several examinations.
Fig. 3. Energy-dispersive X-ray analysis (EDXA) of crystals produced by *A. niger* and *P. javanicus* during growth in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA-amended MCD medium. (A,C) Lead-containing biomineral produced by *A. niger* (shown in Fig. 3B,E). (B,D) Lead-containing biomineral produced by *P. javanicus* (shown in Fig. 3C,F). Typical spectra are shown from one of at least three determinations.

Fig. 4. Transmission electron microscopy (TEM) of biominerals formed by *A. niger* and *P. javanicus* after growth in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P-amended MCD liquid medium. (A,B,C) Biominerals formed in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P-amended MCD liquid medium after growth of *A. niger* for 30 d. Scale bars: A = 3 µm, B,C = 6 µm. (D,E,F) Biominerals formed in 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P-amended MCD liquid medium after growth of *P. javanicus* for 30 d. Scale bars: D = 7 µm, E = 4 µm, F = 2 µm. All organisms were grown for 30 d at 25°C in the dark on an orbital shaking incubator at 125 rpm. Typical images are shown from several examinations.

Fig. 5. X-ray powder diffraction (XRPD) patterns of biominerals extracted from 5 mM Pb(NO$_3$)$_2$ and 30 mM G2P or 5 mM PyA-amended MCD medium after growth of (A) *A. niger* and (B) *P. javanicus*. Patterns for dominant mineralogical components are shown, as well as the new biominerals produced as a result of fungal activity, lead oxalate (PbC$_2$O$_4$) and pyromorphite, syn (Pb$_5$(PO$_4$)$_3$Cl). Mineral particulates were harvested after growth for 30 days at 25°C in the dark on an orbital shaking incubator at 125 rpm. Typical patterns are shown from one of several determinations.
Fig. 6. Predominance diagrams of chemical and mineralogical species of lead. (A) The stability field of pyromorphite in the system Pb, Mg, S, K, Cl, N, C, Na, Fe, P, H and O. (B) The stability field of lead oxalate in the system Pb, Mg, S, K, Cl, N, C, Na, Fe, P, H and O. (C) Lead mineral stability diagrams pH vs log oxalate concentration. Total concentrations for Pb and P in the system were assumed to be 5 and 30 mM respectively, and 30 mM for oxalate, and the concentrations of other elements were taken to be the same values as present in MCD medium.