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Micromolar Intracellular Hydrogen Peroxide Disrupts Metabolism by Damaging Iron-Sulfur Enzymes*

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An *Escherichia coli* strain that cannot scavenge hydrogen peroxide has been used to identify the cell processes that are most sensitive to this oxidant. Low micromolar concentrations of H_2O_2 completely blocked the biosynthesis of leucine. The defect was tracked to the inactivation of isopropylmalate isomerase. This enzyme belongs to a family of [4Fe-4S] dehydratases that are notoriously sensitive to univalent oxidation, and experiments confirmed that other members were also inactivated. *In vitro* and *in vivo* analyses showed that H_2O_2 directly oxidized their solvent-exposed clusters in a Fenton-like reaction. The oxidized cluster then degraded to a catalytically inactive [3Fe-4S] form. Experiments indicated that H_2O_2 accepted two consecutive electrons during the oxidation event. As a consequence, hydroxyl radicals were not released; the polypeptide was undamaged; and the enzyme was competent for reactivation by repair processes. Strikingly, in scavenger-deficient mutants, the H_2O_2 that was generated as an adventitious by-product of metabolism (<1 μM) was sufficient to damage these [4Fe-4S] enzymes. This result demonstrates that aerobic organisms must synthesize H_2O_2 scavengers to avoid poisoning their own pathways. The extreme vulnerability of these enzymes may explain why many organisms, including mammals, deploy H_2O_2 to suppress microbial growth.

Virtually all organisms express peroxidases and catalases to protect themselves from hydrogen peroxide. H_2O_2 is continuously formed by the autoxidation of redox enzymes (reviewed in Ref. 1), and scavenging enzymes may have originally evolved to protect cells against these internal sources of H_2O_2. The peroxidases and catalases are sufficiently abundant and active that they probably drive the steady-state level of intracellular H_2O_2 into the low nanomolar range (2). Nevertheless, it is widely suspected that even this dose of H_2O_2 may compose a chronic, low level stress that gradually debilitates cells and, in higher organisms, drives the deterioration of tissue function as part of the aging process.

Exogenous H_2O_2 rapidly diffuses across cell membranes (2) and can impose a much more acute stress on cells; accordingly, it is often used as a biological weapon. For example, H_2O_2 is formed by phagocytes and may accumulate to 10^{-4} M inside phagosomes that have engulfed invading bacteria. Lactic acid bacteria suppress the growth of competitors by releasing H_2O_2 as a primary metabolic product, achieving millimolar concentrations in laboratory cultures. And redox-cycling antibiotics, which are produced as microbicides by both plants and bacteria, suffuse target organisms with a continuous stream of H_2O_2.

If we wish to understand the severity and nature of the stress that H_2O_2 imposes upon cells, we must identify the biomolecules with which it primarily reacts. This problem has not been easy to solve. *In vitro* studies have shown that H_2O_2 can oxidize methionine (3) and cysteine (4) residues, but the rates at which it does so suggest that these types of damage will be scant at physiological doses of H_2O_2, unless the surrounding polypeptide context somehow strongly activates the residues. Reactions between H_2O_2 and loosely bound iron generate hydroxyl radicals (Reaction 1) and are suspected of being involved in protein carbonylation, lipid peroxidation, and DNA oxidation (5).

$$Fe^{2+} + H_2O_2 \rightarrow HO^{\bullet} + OH^{-} + Fe^{3+}$$

REACTION 1

Early measurements indicated that this reaction (the Fenton reaction) is relatively slow as well (6), prompting some workers to question its significance in real-world scenarios (discussed in Ref. 5); however, subsequent work revealed that anionic ligands activate ferrous iron to the point that it reacts quickly with micromolar H_2O_2 (7, 8).

An alternative approach to pinpointing the important targets of H_2O_2 is to expose cells to increasing doses in a way that identifies the first cell processes to fail. *Escherichia coli* and other organisms have calibrated their defensive systems to detect submicromolar levels of H_2O_2 (2, 9), so we anticipate that these low concentrations are sufficient to threaten the most sensitive biomolecules. Unfortunately, if scavenging enzymes are active, it is difficult to impose such a low dose of H_2O_2 over an extended period of time because the enzymes will degrade the H_2O_2 and end the stress. Therefore, these experiments are most easily conducted with scavenger-deficient mutants. We have constructed *E. coli* strains that lack peroxidase and catalase activities (10). These mutants grow at wild-type rates in anaerobic environments, but when they are exposed to oxygen, they grow at reduced rates in complex medium and fail to grow at all in minimal medium. The former defect is due, at least in part, to Fenton-mediated DNA damage (8). The second defect stems from problems with biosynthetic pathways. In this study, we identified the mechanism by which micromolar H_2O_2 blocks

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TABLE 1
Strains and plasmids used in this study
FRT, Flp recognition target.

	Genotype/characteristics	Source/Ref.
Strain		
MG1655	Wild-type *E. coli*	45
LC106	Δ(*katG17*::Tn*10*)*1* Δ(*ahpC-ahpF'*) *kan*::'*ahpF* Δ(*katE12*::Tn*10*)*1*	10
JH400	Δ(*dgs-fumCA-manA*)*1* Δ(*mel-fumB*) *zdg232*::Tn*10 metB1 spoT1 relA*	John Guest
OD570	Δ*fumC*::*cm*	19
OD571	Δ*fumCA*::*cm*	19
SJ19	Δ*fumC*::*cm* in LC106	This study
SJ20	Δ*fumAC*::*cm* in LC106	This study
SJ34	Δ*fumB*::*cm* in BW25113	This study
SJ37	As LC106 plus Δ*fumC* Δ*fumB*::*cm*	This study
SJ38	As LC106 plus Δ*fumAC* Δ*fumB*::*cm*	This study
BW25113	$lacI^q$ $rrnB_{T14}$ Δ$lacZ_{WJ16}$ *hsdR514* Δ$araBAD_{AH33}$ Δ$rhaBAD_{LD78}$	12
Plasmid		
pWKS30	P_{lac} polylinker Am^r	18
pLEUCD2	pWKS30 containing *leuCD* insert	This study
pCKR101	P_{lac}-$lacI^q$ P_{tac} polylinker Am^r	Jeff Gardner
pLEUCD3	pCKR101 containing *leuCD* insert	This study
pFUMA	pCKR101 containing *fumA* insert	This study
pCP20	Temperature-sensitive Flp expression plasmid	46
pKD3	FRT-flanked *cat*	12
pKD46	λ-Red recombinase expression plasmid	12

leucine biosynthesis, and we found that this class of injury affects multiple pathways in the cell.

EXPERIMENTAL PROCEDURES

Strains and Culture Conditions—The strains and plasmids used in this study are listed in Table 1. Anaerobic cultures were grown in an anaerobic chamber (Coy Laboratory Products Inc.), and aerobic cultures were grown with vigorous shaking in a water bath at 37 °C. Standard minimal medium contained minimal A salts (11), 0.2% glucose, 1 mM $MgCl_2$, 5 mg/liter thiamine, 0.5 mM histidine, 0.5 mM phenylalanine, 0.5 mM tyrosine, and 0.5 mM tryptophan. Histidine was always added to the media because the parent strain, MG1655, is a histidine auxotroph anaerobically; to minimize the difference between anaerobic and aerobic cultures, histidine was also added to aerobic cultures. Where indicated, lactose (0.2%), gluconate (0.2%), or malate (40 mM) was substituted for glucose. Where indicated, other amino acids, α-ketoisovalerate (TCI America) and α-ketoisocaproate (Sigma), were present at 0.5 mM.

Mutations were introduced into new strains by P1 transduction (11). To create a null mutation of *fumB* using the Red recombinase method (12), the forward primer 5′-TAACAAATACAGAGTTACAGGCTGGAAGCTGTAGGCTGGAGCTGC and the reverse primer 5′-AGCATGCTGCCAGGCGCTGGGCCGAAGAGGATATGAATATCCTCC were used. Mutations were confirmed by PCR analysis and enzyme assays.

Aerobic Cell Growth—To ensure that cells were growing exponentially before they were exposed to oxygen, anaerobic overnight cultures of hydroperoxidase-deficient (Hpx^-)[2] cells were diluted to $A_{600} = 0.005$ in fresh anaerobic glucose minimal medium. Cells were then grown anaerobically to $A_{600} \sim 0.1$ prior to dilution into aerobic medium.

Enzyme Assays—Cell extracts were prepared by suspending and sonicating cells in anaerobic buffers inside an anaerobic chamber. Isopropylmalate isomerase (IPMI) activity was measured by monitoring the decrease in the absorbance (235 nm) of citraconate (Sigma) (13), an analog of isopropylmalate; reactions contained 100 mM Tris-Cl (pH 7.6) and 0.4 mM citraconate. Fumarase activity was determined from the appearance of fumarate (250 nm) in a reaction containing 50 mM sodium phosphate (pH 7.4) and 50 mM malate (Sigma) (14). To assay 6-phosphogluconate dehydratase, lysates were prepared from cultures grown in minimal medium containing 0.2% gluconate. Turnover of 6-phosphogluconate dehydratase produces pyruvate; its formation (in 50 mM Tris-Cl (pH 7.65)) was determined in a second reaction catalyzed by lactate dehydrogenase (Sigma) (15). To assay NADH dehydrogenase I, inverted membrane vesicles were isolated from extracts after sonication in 50 mM MES (pH 6.0) (16). NADH dehydrogenase I oxidizes deamino-NADH (340 nm) in 50 mM potassium phosphate buffer (pH 7.8), whereas NADH dehydrogenase II does not. Aconitase was assayed by the conversion of isocitrate to aconitate (17).

Plasmid Construction—The *leuCD* open reading frame was PCR-amplified from *E. coli* MG1655 using the forward primer 5′-ATATCGAATTCTTAACGATATCATTGCCCGCTATATAGCAG-3′ and the reverse primer 5′-CTGGATCTAGATTAATTCATAAACGCAGGTTG-3′. To construct the pLEUCD2 plasmid, the PCR products were digested with XbaI and EcoRI and cloned into the pWKS30 vector (18) behind the *lac* promoter. The plasmid was confirmed by restriction analysis and IPMI assay. Hpx^- cells containing pLEUCD2 or pWKS30 were cultured in lactose minimal medium with histidine, aromatic amino acids, and 50 μg/ml ampicillin. This plasmid was used in complementation experiments; the *leuCD* genes were induced 3-fold above wild-type levels by 1 mM isopropyl β-D-thiogalactopyranoside, which was added to both anaerobic precultures and final aerobic cultures.

For EPR experiments and purification purposes, IPMI and fumarase A were strongly overproduced by expression from a *tac* promoter. The *leuCD* genes were excised from pLEUCD2 with XbaI and EcoRI and cloned into the pCKR101 vector to construct pLEUCD3. The *fumA* open

[2] The abbreviations used are: Hpx^-, hydroperoxidase-deficient; IPMI, isopropylmalate isomerase; MES, 4-morpholineethanesulfonic acid.

reading frame was PCR-amplified from *E. coli* MG1655 using the forward primer 5′-ATATCGAATTCTTAACATAACCAAACCAGGCAGTAAGTG-3′ and the reverse primer 5′-CATGGATCTAGATTATTTCACACAGCGGGTGCATTG-3′. PCR products were digested with XbaI and EcoRI and cloned into the pCRK101 vector to construct pFUMA. Cells containing the plasmids were cultured in glucose minimal medium with histidine, aromatic amino acids, and 50 μg/ml ampicillin. These plasmids overproduced IPMI and fumarase A >60-fold above wild-type levels.

Inactivation of Enzymes—Hpx$^-$ cells were grown anaerobically to A_{600} = 0.2. H_2O_2 was added to the cultures when they were aerated. At designated time points, aliquots were removed; catalase was added to 200 units/ml; and cells were returned to the anaerobic chamber for lysis and assay.

In vivo inactivation of IPMI by endogenous H_2O_2 was initiated by aerating heretofore anaerobic cultures without any addition of exogenous H_2O_2. Under these conditions, cells steadily generate H_2O_2. The H_2O_2 equilibrates so quickly across membranes that, in Hpx$^-$ cultures, the intracellular H_2O_2 concentration is essentially equivalent to the extracellular concentration (2). Extracellular H_2O_2 was measured directly by the Amplex Red/horseradish peroxidase method (2). At the same time, extracts were prepared and assayed in the anaerobic chamber.

Inactivation of enzymes *in vitro* was accomplished by the addition of H_2O_2 to lysates or to purified enzyme in anaerobic buffer. The H_2O_2 was subsequently removed by catalase prior to anaerobic assay. In some cases, damaged iron-sulfur clusters were chemically rebuilt by incubation with 50 μM $Fe(NH_4)_2(SO_4)_2$ (Sigma) and 2.5 mM dithiothreitol (Sigma) (17) at room temperature.

EPR Analysis—In vivo EPR samples were prepared with Hpx$^-$ cells overproducing IPMI or fumarase A. To overexpress the structural genes, 1 mM isopropyl β-D-thiogalactopyranoside was added when the cells reached A_{600} = 0.2. After another 2 h of incubation, the cells were harvested by centrifugation, and the cell pellets were resuspended at 1/500th of the original culture volume in 10% glycerol. The resuspended cells were incubated with H_2O_2 for 1 min at 37 °C. The cell suspension (250 μl) was then transferred into an EPR tube and frozen in dry ice. EPR spectra of $[3Fe\text{-}4S]^+$ clusters (19) were obtained at the following settings: microwave power, 1 milliwatt; microwave frequency, 9.05 GHz; modulation amplitude, 8 G at 100 KHz; and time constant, 0.032.

Purification of Enzymes—Four-liter cultures of the Hpx$^-$ mutant overproducing fumarase A were grown anaerobically to A_{600} = 0.2 at 37 °C. Expression of *fumA* was induced by incubation with 1 mM isopropyl β-D-thiogalactopyranoside for 2 h at 37 °C. The cultures were harvested by centrifugation; the cell pellets were resuspended in 15 ml of anaerobic 50 mM Tris-Cl and 10 mM Mg^{2+} (pH 8) and disrupted by sonication; and cell debris was removed by centrifugation. The purification procedure was derived from the protocol described by Flint *et al.* (20). All steps of the purification were conducted in an anaerobic chamber at room temperature, and all buffers were anaerobic. Protamine sulfate (1%) was added to the supernatant to remove nucleic acids. The supernatant was then loaded onto a DEAE-Sepharose column (16 × 140 mm) and eluted by a 0–1 M gradient of KCl in 50 mM Tris-Cl and 10 mM Mg^{2+}. The fractions containing the highest activity were pooled. Ammonium sulfate (1.5 M) was added; the precipitate was removed by centrifugation; and the supernatant was loaded onto a phenyl-Sepharose column (16 × 110 mm). The fractions were eluted by a 1.5 to 0 M gradient of ammonium sulfate in 50 mM Tris-Cl and 10 mM Mg^{2+}. The fractions containing the highest activity were pooled and concentrated (Amicon). The enzyme was loaded onto a Superdex column (16 × 50 mm). The fractions were eluted with 500 mM KCl in 50 mM Tris-Cl and 10 mM Mg^{2+}. The three fractions containing the highest activity were frozen in dry ice containing ethanol. Purified fumarase A was >90% pure based on SDS-PAGE. The enzyme was quantified by dye binding assay using bovine serum albumin as a standard and the conversion factor determined by Flint *et al.* (20). Aconitase A was overproduced and purified as described (17).

Detection of Iron Released upon Cluster Oxidation—EPR spectroscopy was used to quantitatively correlate the creation of $[3Fe\text{-}4S]^+$ clusters with the release of iron when fumarase A was damaged *in vitro*. Desferrioxamine (1 mM; Sigma) was added to purified fumarase A (18 μM) immediately prior to the addition of H_2O_2. The sample was then transferred into an EPR tube and frozen. The $[3Fe\text{-}4S]^+$ signal was detected as described above, whereas EPR spectra of free iron were obtained at the following settings: microwave power, 10 milliwatt; microwave frequency, 9.05 GHz; modulation amplitude, 12.5 G at 100 KHz; and time constant, 0.032. Iron concentrations were quantified using standard solutions of $FeCl_3$ (Sigma) in 50 mM Tris-Cl containing 10 mM Mg^{2+} and 1 mM desferrioxamine. Desferrioxamine binds both ferric and ferrous iron and triggers the oxidation of the latter species. Thus, the EPR method did not distinguish whether the released iron was in the ferric or ferrous form.

Ferene is commonly used to detect ferrous iron released by metalloenzymes, but we found that ferene itself directly inactivated fumarase A. Dipyridyl did not. Dipyridyl binds ferrous iron in a complex that exhibits an absorbance maximum at 522 nm (21); we determined that this complex cannot be oxidized by H_2O_2. To detect Fe^{2+} released during cluster decomposition, 1 mM dipyridyl (Sigma) was added to 18 μM fumarase A prior to H_2O_2 addition. Control experiments confirmed that this concentration of dipyridyl captured ferrous iron before H_2O_2 could oxidize it: when $Fe(NH_4)_2(SO_4)_2$ was added to the H_2O_2-containing reaction mixture, we were able to quantitatively recover it as a dipyridyl chelate, and we could accurately detect as little as 2 μM.

Protein Mass Spectroscopy—Fumarase A (3 μM) was treated with 5 μM H_2O_2 for 2 min at room temperature. The reaction was terminated by the addition of 0.1 unit/μl catalase. Assays showed that >98% of the enzyme had been inactivated. For mass spectroscopic analysis, 2 μg of sample was desalted using the Genotech Perfect-FOCUS two-dimensional sample cleanup kit. The desalted sample was suspended in 25 mM ammonium bicarbonate containing 12.5 μg/ml trypsin and incubated for 12 h at 37 °C. The sample was then dried, suspended in 5% acetonitrile and 0.1% formic acid, and injected into a Waters Q-Tof quadrupole time-of-flight mass spectrometer via a high pressure liquid chromatography interface. Pep-

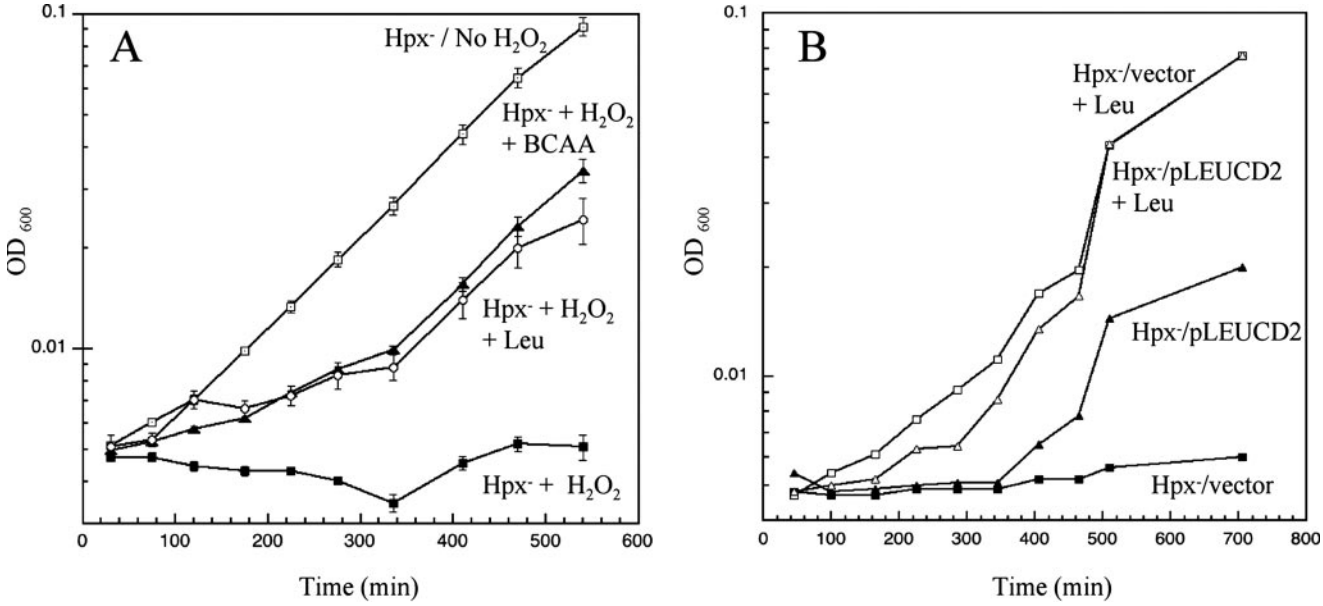

FIGURE 1. **Low concentrations of H_2O_2 block leucine biosynthesis.** *A*, Hpx⁻ cells were cultured in aerobic glucose medium. Where indicated, 8 μM H_2O_2, branched-chain amino acids (*BCAA*; Leu, Ile, and Val), and/or leucine was added. *B*, Hpx⁻ cells containing either pLEUCD2 (overproducing IPMI) or pWKS30 (an empty vector) were cultured in lactose minimal medium with or without leucine supplementation. All media included histidine and aromatic amino acids. The pLEUCD2 plasmid boosted the IPMI activity of anaerobic cultures ~3-fold.

tide masses were detected using a data-dependent method and were subjected to tandem mass spectrometry for *de novo* sequencing and characterization of post-translational modifications. Analysis was done using ProteinLynx Global SERVER Version 2.1 (Waters), Mascot (Matrix Science), and PEAKS (Bioinformatics Solutions Inc.).

RESULTS

Hydroperoxidase mutants that cannot scavenge H_2O_2 (*katG katE ahp*; here denoted Hpx⁻) grow well in anaerobic glucose minimal medium but stop growing when they are aerated. The cells resume growth when aromatic amino acids are supplied, indicating that endogenously formed H_2O_2 poisons some step in the aromatic biosynthetic pathway. The mechanism is unknown and is the subject of a separate investigation. However, aromatic supplements do not fully restore these mutants to a wild-type growth rate, and the addition of small amounts of exogenous H_2O_2 exacerbates the residual defect. Eight micromolar H_2O_2 completely blocked growth in this medium (Fig. 1*A*). Supplementation with casamino acids restored growth, implying that stasis was a result of a second amino acid biosynthetic defect. Experiments using different amino acids determined that leucine was the critical one.

Hpx⁻ growth in the presence of H_2O_2 was improved by supplementation with exogenous α-ketoisocaproate, the final intermediate in the leucine pathway, but not by α-ketoisovalerate, the first one (data not shown). There are five reactions between the two intermediates catalyzed by four enzymes. We anticipated that H_2O_2 might inhibit or inactivate one of them. Indeed, the growth defect was partially relieved by a plasmid that overproduces IPMI 3-fold (Fig. 1*B*), suggesting that this is the rate-limiting enzyme in the pathway during H_2O_2 stress.

Enzyme assays confirmed that IPMI lost activity rapidly when 8 μM H_2O_2 was added to cultures (Fig. 2*A*). Furthermore, significant enzyme inactivation occurred upon aeration even without the addition of exogenous H_2O_2 (Fig. 2*B*), indicating that <0.4 μM intracellular H_2O_2 was sufficient to poison a substantial fraction of IPMI. Thus, this enzyme is exquisitely sensitive, and scavenging enzymes are needed to protect it from endogenously formed H_2O_2.

The Nature of IPMI Damage—IPMI activity was lost when H_2O_2 was added to anaerobically prepared Hpx⁻ extracts (Fig. 3*A*), indicating that inactivation occurred by direct action of H_2O_2 upon the enzyme. Catalase protected completely (data not shown).

IPMI is a dehydratase, and its protein sequence suggests that it belongs to a family of enzymes that employ [4Fe-4S] clusters as active-site catalysts (22). The solvent-exposed clusters of these enzymes both coordinate substrate and act as Lewis acids in abstracting the hydroxide anion from the bound substrate (23). These enzymes are typified by aconitase, and they are notoriously sensitive to inactivation by univalent oxidants such as superoxide and ferricyanide (15, 24–27). These agents abstract a single electron from the cluster, converting it to a $[4Fe\text{-}4S]^{3+}$ form. The cluster is unstable at that valence and releases the substrate-binding iron atom as Fe^{2+} so that the residual cluster is left in a $[3Fe\text{-}4S]^{+}$ form that lacks the key iron atom and is catalytically inactive.

Consistent with this model, the addition of citraconate, a pseudosubstrate of IPMI, protected the enzyme from H_2O_2 *in vitro*, suggesting that H_2O_2 must directly contact the cluster to inactivate the enzyme (data not shown). Damaged clusters can often be reassembled chemically by treatment with dithiothreitol and ferrous iron; when damaged IPMI

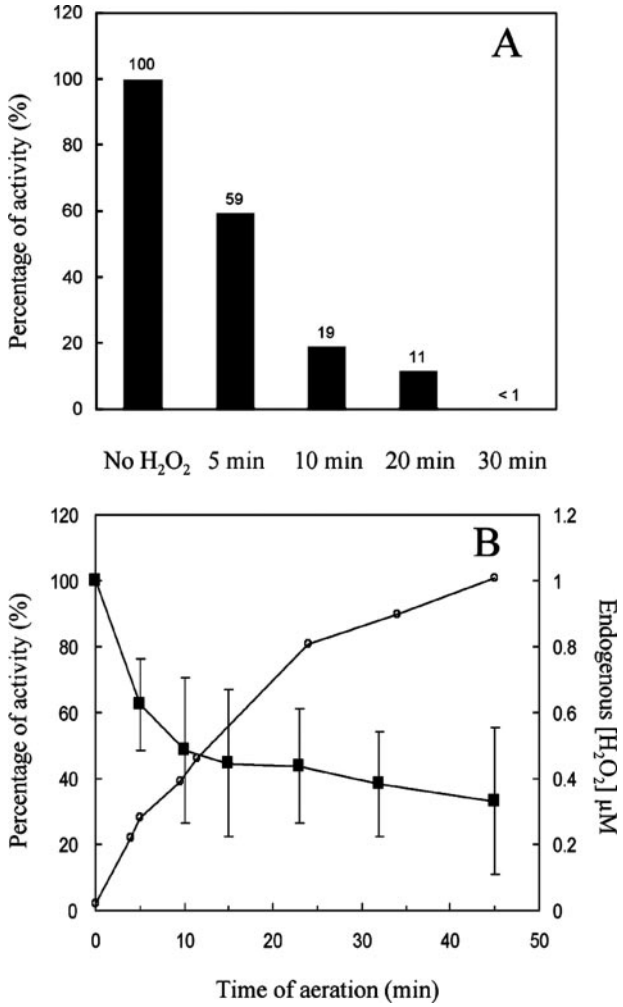

FIGURE 2. **H_2O_2 rapidly inactivates IPMI *in vivo*.** *A*, Hpx$^-$ cells were anaerobically cultured to A_{600} = 0.2, and 8 μM H_2O_2 was then added to the cultures. Catalase was added at intervals, and residual enzyme activity was determined. *B*, Hpx$^-$ cells were cultured in anaerobic medium and then aerated starting at time 0. IPMI activity (■) and H_2O_2 concentrations (○) were monitored.

was subjected to this protocol, 60% of the activity was regained within 3 min (Fig. 3*B*).

IPMI was overexpressed in Hpx$^-$ cells, and cells were then exposed to H_2O_2. A strong [3Fe-4S]$^+$ signal appeared (Fig. 3*C*), which was absent in non-overproducing controls. This result confirmed both that IPMI has a cluster and that it is destroyed by H_2O_2. In fact, ~85% of the IPMI activity was recovered when the extracts from H_2O_2-exposed cells were treated with dithiothreitol and ferrous iron.

Other [4Fe-4S] Dehydratases Are Similarly Sensitive to H_2O_2—Other cluster-containing dehydratases (6-phosphogluconate dehydratase and fumarases A and B) also lost activity when they were exposed to low concentrations of H_2O_2 *in vitro* (Fig. 4, *A*–*C*). In each case, full activity could be restored by subsequent treatment with dithiothreitol and ferrous iron (data not shown). Furthermore, a strong [3Fe-4S]$^+$ signal was detected when H_2O_2 was added to Hpx$^-$ cells overproducing fumarase A (Fig. 5). Thus, it appears that H_2O_2 efficiently damages the clusters of all members of this enzyme family.

Glucose medium, which we employed for our initial growth experiments, does not demand that *E. coli* process substrate through its tricarboxylic acid cycle to generate ATP; therefore, growth would not have been affected by the inactivation of aconitase or fumarase. However, the Hpx$^-$ strain exhibited a severe growth defect when it was cultured in malate medium (Fig. 6). This result indicates that the submicromolar H_2O_2 that is generated by endogenous processes is sufficient to debilitate this pathway.

Iron-sulfur clusters are also used by respiratory enzymes to transfer electrons between active sites. The NADH dehydrogenase I complex is characteristic of these enzymes, as it contains at least nine iron-sulfur clusters (28). However, even 5 mM H_2O_2 was unable to diminish its activity (data not shown). Because these clusters are buried within polypeptide, the implication is that H_2O_2 can oxidize only those clusters that it can contact directly.

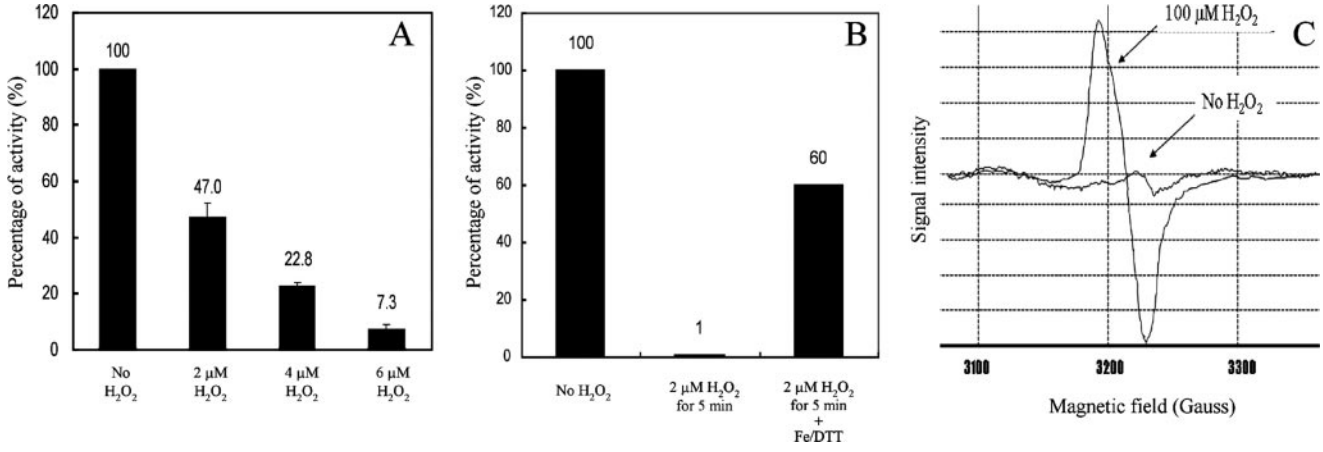

FIGURE 3. **H_2O_2 inactivates IPMI by converting its [4Fe-4S]$^{2+}$ cluster to a [3Fe-4S]$^+$ cluster *in vitro*.** *A*, an extract of anaerobically grown Hpx$^-$ cells was exposed to the indicated concentrations of H_2O_2 for 2 min at room temperature. Catalase was then added to remove the H_2O_2. The experiment was conducted anaerobically. *B*, IPMI from anaerobically cultured Hpx$^-$ cells was inactivated with 2 μM H_2O_2 for 5 min. Catalase was added to remove the H_2O_2, and 50 μM $Fe(NH_4)_2(SO_4)_2$ (*Fe*) and 2.5 mM dithiothreitol (*DTT*) were then added. The activity was measured after 3 min of incubation. *C*, Hpx$^-$ cells overproducing IPMI were harvested when the cells reached A_{600} = 0.2. The cell pellets were resuspended at $^1/_{500}$th of the original culture volume in 10% glycerol. The resuspended cells were incubated with 100 μM H_2O_2 for 1 min at 37 °C. The cell suspension (250 μl) was then transferred into an EPR tube and frozen.

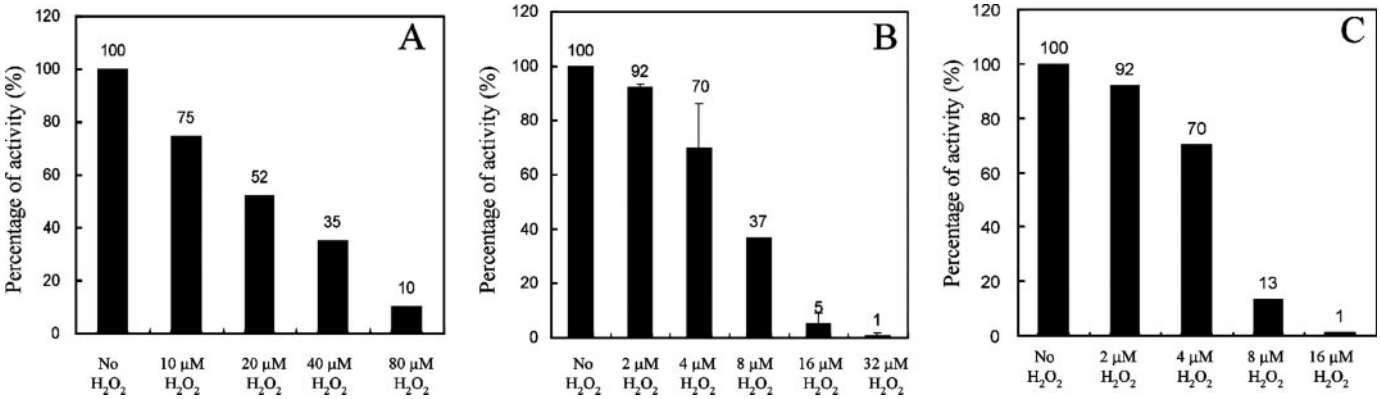

FIGURE 4. **H_2O_2 inactivates other [4Fe-4S] dehydratases.** *A*, 6-phosphogluconate dehydratase; *B*, fumarase A; *C*, fumarase B. Lysates were prepared from anaerobic cultures of strains LC106, SJ37, and SJ20, respectively, and the indicated concentrations of H_2O_2 were added for 5 min. Catalase was added to terminate the stress, and residual activity was determined.

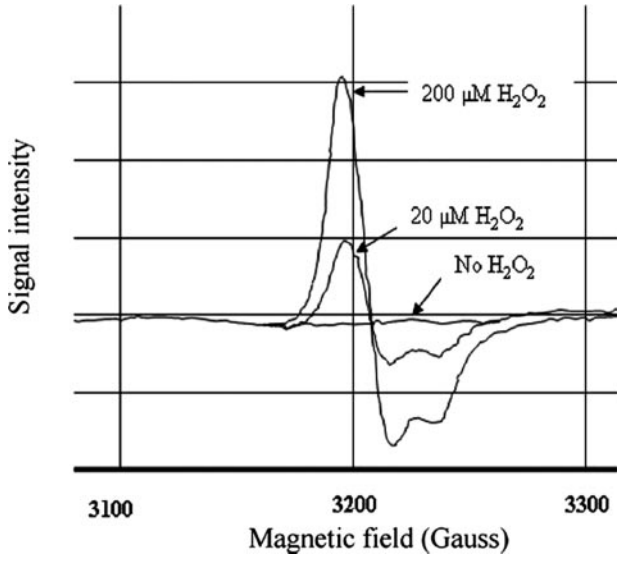

FIGURE 5. **H_2O_2 inactivates fumarase A by converting its $[4Fe\text{-}4S]^{2+}$ cluster to a $[3Fe\text{-}4S]^{+}$ cluster *in vivo*.** SJ37 cells overproducing fumarase A were harvested at A_{600} = 0.2, washed, and resuspended at 1/500th of the original culture volume in 10% glycerol. The resuspended cells were incubated with 200 or 20 μM H_2O_2 for 1 min at 37 °C. The cell suspension (250 μl) was then transferred into an EPR tube and frozen.

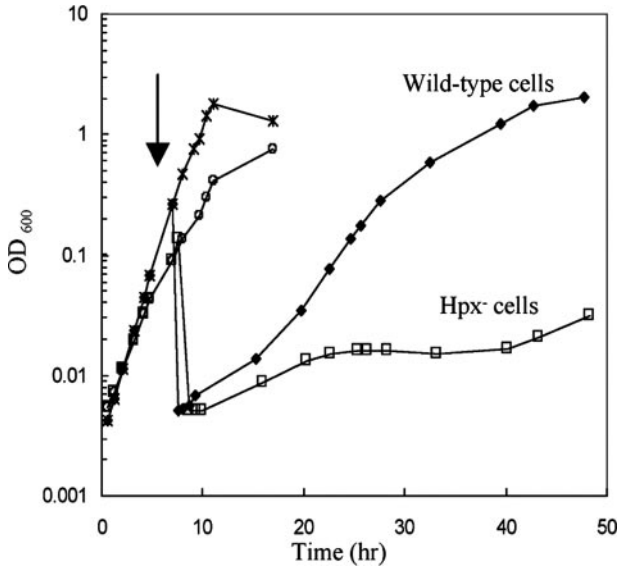

FIGURE 6. **The Hpx⁻ mutant is defective in using malate as the sole carbon source.** Wild-type (*) and Hpx⁻ (○) cells were cultured in aerobic glucose medium to A_{600} = 0.15–0.25. At the *arrow*, aliquots were removed, washed, and resuspended in aerobic malate medium (wild-type (◆) and Hpx⁻ (□) cells). All media included histidine and aromatic amino acids.

The Mechanism of Cluster Inactivation—IPMI is a two-subunit enzyme that dissociates during purification. Therefore, fumarase A and aconitase A were chosen for purification and further examination. The isolated enzymes were acutely sensitive to H_2O_2, exhibiting inactivation rate constants of 4×10^3 and 3×10^2 M^{-1} s^{-1}, respectively, at 0 °C. The fumarase inactivation constant was $0.5–1 \times 10^5$ M^{-1} s^{-1} at 25 °C, and inactivation at 37 °C was too fast for us to measure.

Strikingly, the fumarase rate constants were orders of magnitude higher than the apparent constant that we calculated *in vivo* using Hpx⁻ mutants. We suspected that substrates protect the enzyme inside the cell. Indeed, both malate and fumarate fully protected fumarase when they were added at saturating concentrations (Fig. 7). The doses needed for half-maximal protection (0.4 mM for malate and 0.3 mM for fumarate, measured at 0 °C) were in reasonable agreement with the K_m values of the enzyme for those substrates (0.7 and 0.6 mM, respectively, at 37 °C) (29). Thus, it is likely that the inactivation of the enzyme *in vivo* is tempered by the consequent accumulation of substrate. These data also support the suspicion that H_2O_2 must intimately contact the cluster to inactivate it.

The standard model of univalent cluster oxidation posits that a $[3Fe\text{-}4S]^{+}$ form is initially generated with release of one ferrous ion (27). However, if the oxidant is H_2O_2, then a hydroxyl radical should also be formed (Reactions 2 and 3).

$$[4Fe\text{-}4S]^{2+} + H_2O_2 \rightarrow [4Fe\text{-}4S]^{3+} + OH^- + HO^{\bullet}$$

REACTION 2

$$[4Fe\text{-}4S]^{3+} \rightarrow [3Fe\text{-}4S]^{+} + Fe^{2+}$$

REACTION 3

Hydroxyl radicals react with most organic biomolecules, including amino acids, at nearly diffusion-limited rates; therefore, if one were formed within the active site of a dehydratase, the likely consequence would be the direct oxidation of the protein. However, we were able to quantitatively reac-

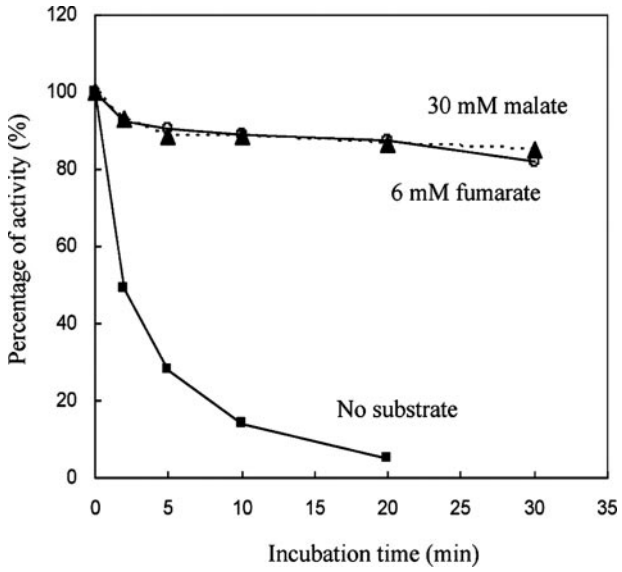

FIGURE 7. **Substrates protect fumarase against H_2O_2.** Purified fumarase A (5 nM) was exposed to 1 μM H_2O_2 at 0 °C. Where indicated, 30 mM malate or 6 mM fumarate was included in the buffer. At designated time points, aliquots were removed; catalase was added; and residual activity was measured.

tivate H_2O_2-treated fumarase (data not shown), which seemed inconsistent with the oxidation of active-site residues. SDS-PAGE analysis of the protein showed that the polypeptide chain was not cleaved. Mass spectroscopy failed to detect any oxidation of histidyl, lysyl, prolyl, or methionyl residues (data not shown).

An alternative model is that a ferryl-like species is generated by the initial electron transfer from the cluster to liganded H_2O_2 and that the ferryl radical then abstracts a second electron from the cluster instead of releasing a hydroxyl radical (Reactions 4 and 5).

$$[4Fe\text{-}4S]^{2+} + H_2O_2 \rightarrow [4Fe\text{-}4S/O]^{2+} + H_2O$$

REACTION 4

$$[4Fe\text{-}4S/O]^{2+} + H^+ \rightarrow [3Fe\text{-}4S]^{+} + Fe^{3+} + OH^-$$

REACTION 5

To test this possibility, we used desferrioxamine and dipyridyl to quantify the release of total iron and ferrous iron, respectively. Ferrous iron is generated by the first mechanistic scheme (Reaction 3), but not by the second (Reaction 6). Although we detected stoichiometric release of one iron atom/cluster (Fig. 8), none of this iron was in the ferrous form (<0.1 atom/cluster) (data not shown). From this result and from the absence of polypeptide oxidation, we infer that a free hydroxyl radical is not generated.

Previous experiments that were conducted with millimolar doses of oxidant indicated that, with time, the cluster disintegration might continue past the $[3Fe\text{-}4S]^+$ state (19). However, the ease with which we were able to chemically reactivate damaged dehydratases was inconsistent with the formation of apoprotein, which was only slowly reactivated. In fact, EPR analysis showed that the $[3Fe\text{-}4S]^+$ fumarase cluster was unaffected by a 10-min exposure to 100 μM H_2O_2 (Fig. 8*A*). We conclude that, over this period, physiological doses of H_2O_2 do not directly degrade clusters beyond the easily repaired $[3Fe\text{-}4S]^+$ state.

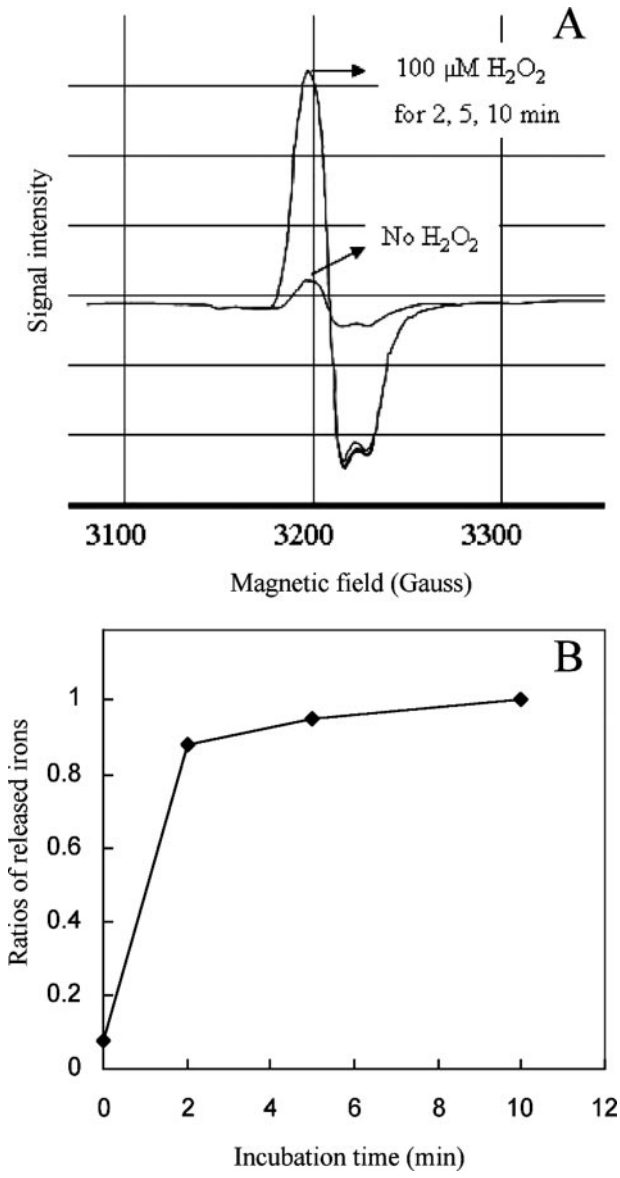

FIGURE 8. **H_2O_2 degrades the $[4Fe\text{-}4S]^{2+}$ cluster of purified fumarase A to a $[3Fe\text{-}4S]^+$ cluster with loss of one iron atom.** Purified fumarase A (18 μM) was inactivated by 100 μM H_2O_2 in 50 mM Tris-Cl and 10 mM Mg^{2+} containing 1 mM desferrioxamine. At designated time points, aliquots were removed; catalase was added; and the reaction mixture was frozen. The residual cluster (*A*) and released ferric iron (*B*) were analyzed by EPR spectroscopy.

DISCUSSION

The experiments reported here reveal that submicromolar concentrations of H_2O_2 are sufficient to destroy enzymatic iron-sulfur clusters. Multiple catabolic and biosynthetic pathways are thereby disrupted. Earlier work with the same strain demonstrated that endogenous H_2O_2 also reacts with unincorporated iron to produce high levels of DNA damage (8). The two phenotypes are of a piece in that both result from Fenton-type reactions.

The sensitivity of iron-sulfur clusters to univalent oxidants has been appreciated for some time; in fact, much of the toxicity of superoxide is due to inactivation of the same enzymes that

were identified in this study. Damage by millimolar doses of H_2O_2 has been reported (17, 30). However, although the chemistry is not unexpected, the surprise is that these reactions occur so rapidly. The rate constant of the Fenton reaction was measured to be 76 M^{-1} s^{-1} at pH 3, and this number has been cited widely (6). It carried with it the implication that reactions between iron and H_2O_2 are too slow to occur at an important rate in most biological scenarios, where H_2O_2 concentrations are micromolar or lower. However, more recent measurements (8) have shown that, at physiological pH, the rate constant for hexa-aqueous iron is actually orders of magnitude higher, presumably because, at neutral pH, hydroxide anion coordinates iron and lowers its reduction potential. Extrapolating from measurements made at lower temperatures (8), we estimate that the rate may be 20,000–30,000 M^{-1} s^{-1} at 37 °C. The same enhancement evidently pertains to the substrate-binding iron atom within a dehydratase iron-sulfur cluster.

The biological consequence of this high reactivity is that far less H_2O_2 is needed to create toxicity than had been suspected. The data directly show that *E. coli* must synthesize scavenging enzymes to avoid being poisoned by the high nanomolar H_2O_2 that it makes through the adventitious oxidation of its own redox enzymes. Basal levels of the NADH peroxidase solve that problem. However, this basic vulnerability is still exploited by phagocytes and plants, both of which suppress the growth of invading bacteria by generating far higher levels of H_2O_2 either directly or through the synthesis of redox-active antibiotics (31–34).

These deliberate oxidative assaults, as well as the H_2O_2 that accumulates in the environment through chemical oxidation processes, are a true threat to bacteria. Because H_2O_2 influx across membranes can outstrip the action of scavenging enzymes, extracellular concentrations of 5 μM H_2O_2 are sufficient to raise the intracellular H_2O_2 in wild-type (scavenger-proficient) cells to ~1 μM (2). Therefore, as little as 5 μM environmental H_2O_2 is sufficient to cause the enzyme inactivity and pathway defects that were reported in this study.

Interestingly, lactic acid bacteria employ a similar stratagem to inhibit competing organisms, utilizing lactate and pyruvate oxidases, which can drive H_2O_2 levels in their immediate environment up to millimolar levels (35, 36). How do the lactic acid bacteria themselves tolerate the stress that they are creating? Lactic acid bacteria do not rely upon pathways that contain iron-sulfur dehydratases. Instead, they obtain from their environment the amino acids that dehydratase-dependent biosynthetic pathways would normally produce, and they ferment sugars to lactic acid rather than using the tricarboxylic acid cycle.

To defend themselves against such H_2O_2-mediated assaults, microbes throughout the biotic world have evolved H_2O_2-inducible defenses, most of which are either homologs or analogs of the OxyR system of *E. coli*. The frontline defense of these systems is the strong induction of scavenging enzymes, but the repair of damaged clusters is also an important feature. Ongoing cluster repair may contribute to the disparity between the rates of enzyme damage that we measured *in vitro* and the apparent rates that we determined *in vivo*. In a previous study (19), we observed that cells quantitatively repaired H_2O_2-damaged dehydratases, but we were perplexed as to why the hydroxyl radical, which presumably had been formed upon cluster oxidation, did not irreversibly damage active-site amino acid residues. This puzzle is resolved by the observation that ferric (rather than ferrous) iron is released during cluster oxidation. Therefore, a ferryl radical (or a non-diffusing hydroxyl radical) evidently pulls a second electron from the residual cluster rather than allowing an unexpended hydroxyl radical to be released into the active-site bulk solution. Interestingly, an analogous phenomenon was noted for the PerR protein of *Bacillus subtilis* in that a Fenton reaction between its prosthetic ferrous iron atom and H_2O_2 caused immediate oxidation of the iron ligands rather than the more widely distributed oxidation of the polypeptide, as might have been expected from a diffusible hydroxyl radical (37).

In the dehydratase case, we do not stipulate whether the second electron transfer would generate an intermediate all-ferric $[4Fe\text{-}4S]^{4+}$ cluster or whether it would create a disulfide bond concomitant with ferric iron release. However, in either case, polypeptide oxidation would be avoided. The physiological significance is that the enzyme can be restored to its native state by cell processes that reduce and remetallate the $[3Fe\text{-}4S]^{+}$ cluster. The components that catalyze that repair process have not yet been identified. However, the OxyR system responds to H_2O_2 by inducing the Suf proteins (38), which compose a backup system for cluster assembly.

A secondary consequence of cluster damage is that iron is released in an uncontrolled manner into the cytosol. Loose iron can bind to DNA and catalyze the formation of damaging hydroxyl radicals; therefore, this iron leak has the effect of accelerating the rate at which H_2O_2 produces DNA damage (39–41). The OxyR system addresses this threat by inducing the synthesis of Dps, a ferritin-like storage protein that scavenges loose iron and sequesters it into an unreactive ferric hydroxide core (8, 42–44). Thus, it has become clear that several aspects of the inducible defense against H_2O_2 are well matched to the threat that H_2O_2 poses.

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REFERENCES

1. Imlay, J. A. (2003) *Annu. Rev. Microbiol.* **57,** 395–418
2. Seaver, L. C., and Imlay, J. A. (2001) *J. Bacteriol.* **183,** 7182–7189
3. Griffiths, S. W., and Cooney, C. L. (2002) *Biochemistry* **41,** 6245–6252
4. Winterbourn, C. C., and Metodiewa, D. (1999) *Free Radic. Biol. Med.* **27,** 322–328
5. Halliwell, B., and Gutteridge, J. M. (1990) *Methods Enzymol.* **186,** 1–85
6. Walling, C. (1975) *Acc. Chem. Res.* **8,** 125–131
7. Rush, J. D., Maskos, Z., and Koppenol, W. H. (1990) *FEBS Lett.* **261,** 121–123
8. Park, S., You, X., and Imlay, J. A. (2005) *Proc. Natl. Acad. Sci. U. S. A.* **102,** 9317–9322
9. Aslund, F., Zheng, M., Beckwith, J., and Storz, G. (1999) *Proc. Natl. Acad. Sci. U. S. A.* **96,** 6161–6165
10. Seaver, L. C., and Imlay, J. A. (2001) *J. Bacteriol.* **183,** 7173–7181

11. Miller, J. H. (1972) *Experiments in Molecular Genetics*, Cold Spring Harbor Laboratory, Cold Spring Harbor, NY
12. Datsenko, K. A., and Wanner, B. L. (2000) *Proc. Natl. Acad. Sci. U. S. A.* **97,** 6640–6645
13. Kohlhaw, G. B. (1988) *Methods Enzymol.* **166,** 423–429
14. Massey, V. (1955) *Methods Enzymol.* **1,** 729–735
15. Gardner, P. R., and Fridovich, I. (1991) *J. Biol. Chem.* **266,** 1478–1483
16. Matsushita, K., Ohnishi, T., and Kaback, H. R. (1987) *Biochemistry* **26,** 7732–7737
17. Varghese, S. M., Tang, Y., and Imlay, J. A. (2003) *J. Bacteriol.* **185,** 221–230
18. Wang, R. F., and Kushner, S. R. (1991) *Gene* (*Amst.*) **100,** 195–199
19. Djaman, O., Outten, F. W., and Imlay, J. A. (2004) *J. Biol. Chem.* **279,** 44590–44599
20. Flint, D. H., Emptage, M. H., and Guest, J. R. (1992) *Biochemistry* **31,** 10331–10337
21. Mason, E. C., and Adarraga-Elizaran, A. (1963) *J. Clin. Pathol.* **16,** 604–606
22. Hentze, M. W., and Argos, P. (1991) *Nucleic Acids Res.* **19,** 1739–1740
23. Lauble, H., Kennedy, M. C., Beinert, H., and Stout, C. D. (1992) *Biochemistry* **31,** 2735–2748
24. Gardner, P. R., and Fridovich, I. (1991) *J. Biol. Chem.* **266,** 19328–19333
25. Kuo, C. F., Mashino, T., and Fridovich, I. (1987) *J. Biol. Chem.* **262,** 4724–4727
26. Liochev, S. I., and Fridovich, I. (1992) *Proc. Natl. Acad. Sci. U. S. A.* **89,** 5892–5896
27. Flint, D. H., Tuminello, J. F., and Emptage, M. H. (1993) *J. Biol. Chem.* **268,** 22369–22376
28. Ohnishi, T. (1998) *Biochim. Biophys. Acta* **1364,** 186–206
29. Flint, D. H. (1994) *Arch. Biochem. Biophys.* **311,** 509–516
30. Brown, N. M., Kennedy, M. C., Antholine, W. E., Eisenstein, R. S., and Walden, W. E. (2002) *J. Biol. Chem.* **277,** 7246–7254
31. Klebanoff, S. J., Locksley, R. M., Jong, E. C., and Rosen, H. (1983) *CIBA Found. Symp.* **99,** 92–112
32. Wojtaszek, P. (1997) *Biochem. J.* **15,** 681–692
33. Inbaraj, J. J., and Chignell, C. F. (2004) *Chem. Res. Toxicol.* **17,** 55–62
34. Ran, H., Hassett, D. J., and Lau, G. W. (2003) *Proc. Natl. Acad. Sci. U. S. A.* **100,** 14315–14320
35. Ito, A., Sato, Y., Kudo, S., Sato, S., Nakajima, H., and Toba, T. (2003) *Curr. Microbiol.* **47,** 231–236
36. Pericone, C. D., Park, S., Imlay, J. A., and Weiser, J. N. (2003) *J. Bacteriol.* **185,** 6815–6825
37. Lee, J. W., and Helmann, J. D. (2006) *Nature* **440,** 363–367
38. Zheng, M., Wang, X., Templeton, L. J., Smulski, D. R., LaRossa, R. A., and Storz, G. (2001) *J. Bacteriol.* **183,** 4562–4570
39. Touati, D., Jacques, M., Tardat, B., Bouchard, L., and Despied, S. (1995) *J. Bacteriol.* **177,** 2305–2314
40. Liochev, S. I., and Fridovich, I. (1994) *Free Radic. Biol. Med.* **16,** 29–33
41. Keyer, K., and Imlay, J. A. (1996) *Proc. Natl. Acad. Sci. U. S. A.* **93,** 13635–13640
42. Almiron, M., Link, A. J., Furlong, D., and Kolter, R. (1992) *Genes Dev.* **6,** 2646–2654
43. Altuvia, S., Almiron, M., Huisman, G., Kolter, R., and Storz, G. (1994) *Mol. Microbiol.* **13,** 265–272
44. Zhao, G., Ceci, P., Ilari, A., Giangiacomo, L., Laue, T. M., Chiancone, E., and Chasteen, N. D. (2002) *J. Biol. Chem.* **277,** 27689–27696
45. Neidhardt, F. C., Curtiss, R., III, Ingraham, J. L., Lin, E. C. C., Low, K. B., Magasanik, B., Reznikoff, W. S., Riley, M., Schaechter, M., and Umbarger, H. E. (1996) in *Escherichia coli and Salmonella* (Neidhardt, F. C., ed) ASM Press, p. 2480, Washington, D. C.
46. Cherepanov, P. P., and Wackernagel, W. (1995) *Gene* (*Amst.*) **158,** 9–14

Micromolar Intracellular Hydrogen Peroxide Disrupts Metabolism by Damaging Iron-Sulfur Enzymes

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