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Sodium Selenide Toxicity Is Mediated by O2-Dependent DNA Breaks

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Abstract

Hydrogen selenide is a recurrent metabolite of selenium compounds. However, few studies examined the direct link between this toxic agent and cell death. To address this question, we first screened a systematic collection of Saccharomyces cerevisiae haploid knockout strains for sensitivity to sodium selenide, a donor for hydrogen selenide (H2Se/HSe−/Se2−). Among the genes whose deletion caused hypersensitivity, homologous recombination and DNA damage checkpoint genes were over-represented, suggesting that DNA double-strand breaks are a dominant cause of hydrogen selenide toxicity. Consistent with this hypothesis, treatment of S. cerevisiae cells with sodium selenide triggered G2/M checkpoint activation and induced in vivo chromosome fragmentation. In vitro, sodium selenide directly induced DNA phosphodiester-bond breaks via an O2-dependent reaction. The reaction was inhibited by mannitol, a hydroxyl radical quencher, but not by superoxide dismutase or catalase, strongly suggesting the involvement of hydroxyl radicals and ruling out participations of superoxide anions or hydrogen peroxide. The OH signature could indeed be detected by electron spin resonance upon exposure of a solution of sodium selenide to O2. Finally we showed that, in vivo, toxicity strictly depended on the presence of O2. Therefore, by combining genome-wide and biochemical approaches, we demonstrated that, in yeast cells, hydrogen selenide induces toxic DNA breaks through an O2-dependent radical-based mechanism.

Introduction

Selenium is mainly known as an essential micronutrient of many living species, including humans [1]. At high doses, selenium is poisonous [2]. In the recent years, selenium deserved considerable interest because of its possible protective effect against cancer at subtoxic doses [3]. Beyond its role in chemoprevention, subtoxic administration of selenium appears to also have a promising potential in cancer therapy [2,4,5,6]. In all these medical applications, the gap between toxic and prophylactic or therapeutic doses is narrow. Although recent studies in a variety of model systems have increased our understanding of the anticarcinogenic mechanisms of selenium compounds, efforts still have to be made to complete our basic view of the underlying roles of selenium metabolites [3,7].

Toxicity of selenium possibly combines several mechanisms such as redox cycling, DNA damage, glutathione (GSH) depletion or protein oxidation. Most of these mechanisms are linked to the pro-oxidant properties of some derivatives of selenium. In particular, it was early shown that, in the presence of thiols, selenite (SeO4−2) is reduced to hydrogen selenide, i.e. H2Se, HSe− and Se2−, in chemical equilibrium. Upon oxidation by O2, hydrogen selenide produces elemental selenium and reactive oxygen species (ROS) including superoxide anions (O2−) and hydrogen peroxide (H2O2) [8,9,10]. Production of hydroxyl radicals (OH) was also proposed [11,12], but, to our knowledge, experimental data in favor of such a production were never provided.

Although hydrogen selenide is believed to be a key player in the toxicity of inorganic selenium compounds [2,13], its mechanism of action has hardly been experimentally studied. To fill this gap, we used Saccharomyces cerevisiae as a model [2]. Because this organism lacks the pathway for the specific incorporation of selenocysteine into proteins, interferences with the selenium incorporated in the active site of proteins are precluded.

In S. cerevisiae, several studies dealing with the toxicity of selenite, a precursor of hydrogen selenide, have been reported. A genome-wide transcriptional analysis revealed that selenite promoted the expression of most of the genes involved in the global oxidative stress response [14]. Moreover, glutathione reductase [15,16], glutaredoxins [16,17,18] and Yap1p [16], a key transcriptional activator for the oxidative stress response, were shown to be involved in cell resistance to a selenite stress. All these studies indicate that selenite induces an oxidative stress in vivo. Other
studies pinpoint involvement of selenium derivatives in genotoxic effects including base oxidations [15,19,20] and DNA breaks [15,16,21,22].

To analyze the mechanism of toxicity of hydrogen selenide, we first screened a collection of gene deletion \emph{S. cerevisiae} mutant strains for hypersensitivity to sodium selenide (Na2Se). We found a strong enrichment for homologous recombination (HR) and DNA damage checkpoint genes. Secondly, using flow cytometry, we showed that cells exposed to sodium selenide were blocked in the G2/M cell-cycle phase. Moreover, induction by Na2Se of DNA double-strand breaks (DSBs) \textit{in vivo} was evidenced by pulse-field gel electrophoresis (PFGE) analysis. Next, using supercoiled DNA \textit{in vitro}, we established that free radicals generated in solution by hydrogen selenide in the presence of dioxygen break phosphodiester bonds. Finally, consistent with the idea that the effect on DNA we describe \textit{in vitro} might at least partly reflect what occurs \textit{in vivo}, we observed that dioxygen potentiated the toxicity of hydrogen selenide against \emph{S. cerevisiae} and that a mutant strain defective in HR displayed oxygen-dependent hypersensitivity to selenide.

\section*{Materials and Methods}

\subsection*{Reagents}

Mannitol, glutathione, sodium selenite, 2-morpholinoethanesulfonic acid (MES), 5,5'-dithiobis(2-nitrobenzoic) acid (DTNB), camptothecin (Cpt), dimethyl sulfoxide (DMSO) and propidium iodide were from Sigma. H2O2 solution (30%, w/w) was from Merck. Catalase from beef liver, superoxide dismutase (SOD) from bovine erythrocytes, RNase A from bovine pancreas and Sall restriction enzyme were purchased from Roche Applied Science. Topoisomerase I from \emph{Escherichia coli} was from New England BioLabs. 5-(dithiophosphoryl)-5-methyl-1-pyrroline \(\mathrm{N}\)-oxide (DEPMPO) was obtained from Radical-Vision (Marseille, France). Sodium selenide (Na2Se) was from Alfa Aesar (Bischheim, France). In solution, Na2Se dissociates into 2 Na\(^+\) and Se\(^{2-}\). The latter ion partly protonates to give HSe\(^-\) and H2Se. According to the pK\(_a\) values of HSe\(^-\) (3.9) and HSe\(^{2-}\) (11.0), at pH 6.0 or 7.0, HSe\(^-\) is the most abundant species. For selenide stress challenge experiments, Na2Se powder was dissolved in a deoxygenated 50 mM MES buffer (pH 6.0), in an anaerobic glove box. In the case of flow cytometry, DNA breakage and electron spin resonance (ESR) experiments, dissolution was in a deoxygenated 100 mM potassium phosphate buffer (pH 7.0). Na2Se concentrations in solution were measured using the colorimetric assay described for hydrogen sulfide [23]. Briefly, samples (1 to 100 \(\mu\)l) were mixed with a 100 \(\mu\)M solution of DTNB in 1 ml final volume of MES buffer (50 mM, pH 6.0), prior to measurement of absorbance at 412 nm.

\subsection*{Strains and Media}

The \emph{S. cerevisiae} strain BY4742 (MAT\(a\) his3\(\Delta\)1 leu2\(\Delta\)0 tyr1\(\Delta\)0 ura3\(\Delta\)0) and its isogenic rad52D mutant were from Euroscarf. The RAD52 gene deletion was verified by multiple PCR reactions using primers outside and/or inside the integrated \textit{kanMX} cassette. Rich YPD and YTD media contained 1% yeast extract (Difco), 2% glucose and either 1% Bacto-peptone (Difco) or 1% Bacto-trypton (Difco), respectively. YTD medium was buffered at pH 6.0 by the addition of 50 mM MES-NaOH. Synthetic dextrose (SD) minimal medium contained 0.67% yeast nitrogen base (Difco), 50 mM MES-NaOH (pH 6.0), 2% glucose and 50 \(\mu\)g/ml of each histidine, leucine, lysine and uracil.

To constitute a pool of \emph{S. cerevisiae} mutants, cells from the systematic deletion collection made in strain BY4741 (MAT\(a\) his3\(\Delta\)1 leu2\(\Delta\)0 met15\(\Delta\)0 ura3\(\Delta\)0) [24] were grown in individual wells in 96 deep-well plates at 30\(^\circ\)C for 2 days in YPD medium. Pools of cells were obtained by mixing several cultures, divided in aliquots and stored at \(-80\)^\({\circ}\)C. The aliquots were thawed and allowed to recover in 800 ml of YTD medium for 9 h at 30\(^\circ\)C, under shaking. Next, this preculture was used to inoculate three 1-liter cultures in YTD, at a final optical density (OD) of 0.2 at 650 nm. In two of the cultures, at t = 0, Na2Se was added at a final concentration of either 1 or 2 \(\mu\)M. The third culture was grown in the absence of Na2Se. All three cultures were grown aerobically at 30\(^\circ\)C, under shaking. Every 4 h, the cultures were diluted to an OD\(_{650}\) of 0.2, and, in the case of the two subcultures challenged with selenide, fresh Na2Se was added back at final concentrations of 1 and 2 \(\mu\)M, respectively. At times 0, 16 h and 27 h, 30-ml aliquots were harvested and centrifuged to collect cells.

\subsection*{Microarray Estimation of Growth Rates}

The growth of individual strains in the pool of \emph{S. cerevisiae} mutants was estimated from measurements done on barcode microarrays (see Methods S1 and Table S2 for a detailed protocol). To analyze the data, we calculated a relative fitness defect for each strain, defined as \(r_{f} = \log_{2}(t_{\text{wt}}^{\text{Se}}/t_{\text{mut}}^{\text{Se}}/t_{\text{mut}}^{\text{H}_{2}\text{Se}}(t_{\text{mut}}^{\text{H}_{2}\text{Se}}+1))\), where \(t_{\text{wt}}^{\text{Se}}(t_{\text{mut}}^{\text{Se}})\) and \(t_{\text{mut}}^{\text{H}_{2}\text{Se}}\) are the generation times of the wild-type and mutant strains in the presence (absence) of Na2Se, respectively (see Methods S2).

\subsection*{Flow Cytometry Analysis}

Wild-type BY4742 and its isogenic rad52D mutant were grown at 30\(^\circ\)C in 100 ml YTD liquid medium. When the OD\(_{650}\) reached 0.5, the culture was divided into subcultures of 5 ml each. At t = 0, Na2Se dissolved in phosphate buffer or Cpt solubilized in DMSO were added and incubation was resumed. For the analysis of Cpt effect, DMSO was kept at a final concentration of 2% (v/v) in the growth medium to maintain Cpt solubility. We verified that, in the absence of Cpt, such a concentration of DMSO did not modify the cell-cycle distribution. After 2 h incubation, cells were harvested by centrifugation, washed in 1 ml of phosphate buffer saline (PBS) and fixed in 1 ml of cold 70% (v/v) ethanol. After 3 h at 37\(^\circ\)C in 0.5 ml of PBS containing 1.5 mg/ml of RNase A, propidium iodide was added at a final concentration of 50 \(\mu\)g/ml and cells were incubated for 15 min at room temperature. Finally, cells were resuspended in 0.5 ml of PBS containing 5 \(\mu\)g/ml propidium iodide. Just before analysis with the FACS Calibur (BD Biosciences) cytometer, the solution was briefly sonicated to dissociate cell doublets. Analysis was performed with 60,000 cells for each condition.

\subsection*{PFGE Analysis}

BY4742 cells were grown at 30\(^\circ\)C in 300 ml of SD medium. When the OD\(_{650}\) reached 1.0, the culture was divided in aliquots of 50 ml and sodium selenide was added to each aliquot at a final concentration of 0, 2.5, 5, 10 or 25 \(\mu\)M. After 1 h incubation at 30\(^\circ\)C, cells were harvested by centrifugation, washed 3-times with 1 ml of 0.5 M EDTA (pH 8.0) and finally resuspended in 0.5 ml of 0.05 M EDTA. Chromosomes were prepared from 50 \(\mu\)l of the above cell suspension as described previously [25], except that agarose plugs were additionally incubated overnight at 37\(^\circ\)C with 2 \(\mu\)g/ml of RNase A. Electrophoresis [25] was performed on a CHEF Mapper XA system (Bio-Rad) at 6 V.cm\(^{-1}\) for 28 h at 14\(^\circ\)C. Separation was based on a two-state mode with angles of 60\(^\circ\) and 180\(^\circ\), and a 60–120 s switch time ramp. After electrophoresis, the gel was stained with 50 \(\mu\)g/ml ethidium bromide for 30 min. Images were recorded on a Typhoon 9400 Imager (GE Healthcare) and bands intensities were determined with ImageJ software.
Analysis of DNA Single-strand Breaking in Aerobic Conditions

pNOY102 plasmid [26] (10 μg/ml final concentration) was mixed on ice with potassium phosphate buffer (100 mM, pH 7.0) and with various components in different combinations involving sodium selenide (13 μM), sodium selenite (25 μM), glutathione (500 μM), mannitol (80 mM), catalase (50 units/ml), SOD (100 units/ml), Topo I (200 units/ml) and Sall (500 units/ml). Solutions were incubated 1 h at 37°C. Then, 150 mM of mannitol was added, and tubes were placed on ice. DNA molecules were loaded on a 0.7% (w/v) agarose gel in Tris-borate-EDTA buffer (45 mM Tris-borate (pH 8.3), 1 mM EDTA). After migration, the gel was stained with ethidium bromide.

ESR Spectroscopy

DEPMPO spin trapper stock solution was prepared as previously described [27]. For analysis, solutions (400 μl) contained 250 mM potassium phosphate buffer (pH 7.0) and 120 mM DEPMPO. When added, mannitol, SOD and catalase were used at final concentrations of 150 mM, 100 units/ml and 500 units/ml, respectively. A freshly prepared solution of Na2Se was added to the sample just before analysis. After 5 min of incubation at 37°C, the sample was transferred in the ESR glass cell and spectra were immediately recorded. Control production of hydroxyl radicals by the Fenton reaction was obtained by mixing H2O2 (10 mM), FeSO4 (40 μM) and EDTA (80 μM). ESR experiments were performed with a Bruker EMX spectrometer operating at 9.7 GHz. An aqueous quartz flat-cell was used in a TE102 rectangular cavity. A microwave power of 20 mW and a field modulation of 2 gauss were used. For each sample, 6 successive scans from 338 to 358 mT were recorded (80 s per scan).

Comparison of DNA Single-strand Breaking in Aerobic and Anaerobic Conditions (Glove Box Conditions)

Potassium phosphate (100 mM, pH 7.0) was prepared under aerobic conditions and then split into two aliquots. One of the aliquots was placed for 1 h under argon flow, before its transfer into an oxygen-free glove box (deoxygenated buffer). The other aliquot was introduced into the glove box and resuspended in deoxygenated SD medium. To exhaust the remaining dioxygen, cells were incubated at 30°C for 30 min. Then, they were washed three times with MES buffer (50 mM, pH 6.0) and finally resuspended in MES buffer to obtain a OD650 of 0.1. After 5 min of incubation at 30°C with various concentrations of Na2Se (0–50 μM), cell suspensions were diluted 1000-fold in water and 150 μl of these dilutions were plated in duplicate onto YTD agar plates. Plates were placed inside an anaerobic jar containing a BioMérieux GENbox Anaer small bag. Next, the jar was removed from the glove box to a 30°C incubator. After 5 days, the colonies were counted. For aerobic conditions, the experiment was driven in the same way out of the glove box and with oxygenated solutions. The colonies were counted after a 2-day incubation at 30°C.

Results

A Genetic Screen Highlights a Key Role of Recombinational Repair and Checkpoint Genes in Resistance to Sodium Selenide

To identify cellular processes involved in S. cerevisiae resistance to hydrogen selenide, we analyzed the sensitivity of a collection of approximately 3000 isogenic haploid knockout mutants to sodium selenide, a donor of hydrogen selenide [24]. Because oxidation of hydrogen selenide is rapid in an oxygenated solution (half-life <2 min in rich YTD medium at 30°C and pH 6.0), the selenide stress challenge was performed by renewing sodium selenide addition every 4 h. Two experiments were performed with 1 or 2 μM sodium selenide final concentrations, respectively. At such concentrations, the doubling time of the wild-type strain was increased by 30–40%. Cells were collected after 16 and 27 h. DNA barcode regions of the various strains were amplified by PCR and labeled with Cy3 or Cy5 fluorophores (Figure S1). Hybridization of these PCR products on Agilent barcode microarrays was used to derive relative fitness defect estimates for individual mutants (see Materials and Methods).

As shown in Table S1, the results were similar for the two concentrations of Na2Se used (1 and 2 μM). Thus, the two sets of data were fused. The resulting distribution of the fitness values was asymmetric with a long tail on the negative side, as expected from a selective effect of sodium selenide treatment on a subset of the mutants from the collection (Figure S2A). Because of this asymmetry and of the broader-than-Gaussian shape of the density distribution, a z-type statistics could not be used to analyze the results. Instead, we examined the ranks of the values (see Table 1 for the annotated list of the 30 most selenide-sensitive deletion strains, and Table S1 for the full list).

Two criteria validated the screen a posteriori. First, we found similar sensitivities to sodium selenide when two independent disruptions of a same gene were available and compared. This was the case with overlapping ORFs (Figure S2B, see also, for example, MMS4 and YBR099C, or FIG4 and YNL324W in Table 1). Secondly, deletion mutants for genes coding for distinct subunits of a same protein complex often ranked similarly. This was the case of the Rad55p-Rad57p complex (rank 2 for RAD57 and 45 for RAD55), the Mus81p-Mms4p complex (rank 26 for MUS81 and 52 for MMS4) and the Vac1p-Fig4p complex (rank 29 for VAC1 and 32 for FIG4). Notably, out of the 9 nonessential genes encoding subunits of the Swr1 complex, 7 (SWR1, YAF9, SWC5, ARP5, SWC3, VPS71, VPS72) belonged to the 100 first ranked genes.

Strikingly, our screen revealed that 29% of the 28 genes belonging to the HR pathway ranked in the first 30 (with a p-value
Table 1. Ranking of genes (1 to 30) based on relative fitness defects calculated for deletion strains after treatment with sodium selenide.

<table>
<thead>
<tr>
<th>rank</th>
<th>rfc</th>
<th>gene name</th>
<th>description/function</th>
<th>DSBb</th>
<th>Cptc</th>
<th>γ-raysd</th>
<th>GSH*e</th>
</tr>
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<td>1</td>
<td>−1.75</td>
<td>GSH1</td>
<td>γ-Glutamylcysteine synthetase</td>
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<td></td>
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<td>2</td>
<td>−1.70</td>
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<td>3</td>
<td>−1.68</td>
<td>GLR1</td>
<td>Glutathione reductase</td>
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<td>4</td>
<td>−1.67</td>
<td>HOM6</td>
<td>Homoserine dehydrogenase</td>
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<td></td>
<td></td>
</tr>
<tr>
<td>5</td>
<td>−1.62</td>
<td>YAF9</td>
<td>Chromatin remodeling, DNA repair</td>
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<td></td>
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<td>6</td>
<td>−1.59</td>
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<td>Glutaredoxin 1</td>
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<td>−1.39</td>
<td>RAD9</td>
<td>DNA damage checkpoint</td>
<td></td>
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<td>RAD24</td>
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<td>−1.37</td>
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<td>Nucleotide diphosphatase</td>
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<tr>
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<td>YMR031W-A</td>
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<td>−1.28</td>
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<td>High affinity methionine permease</td>
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<td>DUN1</td>
<td>DNA damage checkpoint</td>
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<td>YNL324W</td>
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<tr>
<td>25</td>
<td>−1.08</td>
<td>SWR1</td>
<td>Chromatin remodeling</td>
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<tr>
<td>26</td>
<td>−1.04</td>
<td>MUS81</td>
<td>Subunit of the Mms4p-Mus81p endonuclease involved in DNA repair</td>
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<tr>
<td>27</td>
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<td>RADS1</td>
<td>Repair of DSBs in DNA</td>
<td>+</td>
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<tr>
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<td>−1.02</td>
<td>VAC14</td>
<td>Synthesis of phosphatidylinositol 3,5-bisphosphate</td>
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<td>−1.02</td>
<td>RPS21B</td>
<td>Protein of the small ribosomal subunit</td>
<td></td>
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</table>

**r**: relative fitness defect.

*DSB*: genes of the list encoding proteins implicated in cellular response to DSBs (HR and DNA damage checkpoint) are labeled with +.

*Cpt*: genes of the list having a low rank in a screen with Cpt, as reported by Hillenmeyer et al. [45]. Genes ranking under 30 are labeled with +, genes ranking between 31 and 150 are labeled with #.

*γ-rays*: genes ranking under 30 in a γ-rays screen, as reported by Game et al. [30] (data regarding higher ranks are no more available on the referenced web site).

*GSH*: the designated proteins are involved in glutathione homeostasis.

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were in the first 250 ranks (median ranks of 1774 and 2505, respectively) (Figure 1). In between, nucleotide excision repair (NER, 10 genes) and post-replicative repair (PRR, 9 genes) showed consistent with DSB being the major DNA damage caused by rad52 activation, both with the wild-type and of the effects of Cpt and sodium selenide on G2/M checkpoint blockage is amplified by 22% and 8%, respectively), indicating that selenide-induced cell-blockage and DNA repair are delayed to G2 [33].

When the wild-type strain was exposed to sodium selenide, the G2/M population increased. At the highest concentration of Na2Se (2 μM), the effect was dramatic. The G2/M population increased by 19% whereas the G1 population dropped to 6%, and quasi-complete synchronization of the cell population occurred. Importantly, at the lowest used concentration of Na2Se (0.5 μM), the increase in the G2/M percentage was nearly three times higher with the rad52Δ strain than with the wild-type strain (22% and 8%, respectively), indicating that selenide-induced cell-cycle blockage is amplified by RAD52 inactivation. The similarity of the effects of Cpt and sodium selenide on G2/M checkpoint activation, both with the wild-type and rad52Δ strains, is consistent with DSB being the major DNA damage caused by selenide.

**Selenium-Induced DNA Double-Strand Breaks**

These results prompted us to determine if DSBs were indeed produced in cells treated with sodium selenide. S. cerevisiae cells grown in minimal medium were incubated in the absence or in the presence of increasing concentrations of Na2Se. After 1 h of incubation, cells were included into agarose plugs and their chromosomes were analyzed by PFGE (Figure 3A). Cell survival after the treatment was also measured (Figure 3B).

In the absence of sodium selenide, yeast chromosomes migrated as discrete bands. In the presence of sodium selenide, the
intensities of the chromosome bands decreased, whereas smears corresponding to shorter DNA fragments accumulated. This DNA fragmentation was accompanied by a decrease in cell viability. To estimate the level of damaged chromosomes in each growth condition, we quantified the bands corresponding to the largest chromosomes (Figure 3B). For smaller chromosomes, bands could not be distinguished from the breakage products of the longer chromosomes. Comparison of the two curves shown in Figure 3B indicates that the increase in chromosome damage was very similar to the variation of cell death. We conclude that, in the presence of Na₂Se in the culture medium, DSBs are produced and that the rate of DSB formation tightly correlates with the rate of cell death.

**Selenide, but not Selenite, Breaks Phosphodiester Bonds in vitro**

The DNA breaks we observed in vivo can be caused by hydrogen selenide either directly or indirectly. To help distinguish between these two possibilities, we asked whether addition of Na₂Se broke DNA in a minimal system consisting of supercoiled DNA in an oxygenated phosphate buffer. In this system, conversion of the supercoiled plasmid into its nicked form which has a lower electrophoretic mobility allows to detect SSBs. Electrophoresis conditions were set up to separate supercoiled, nicked and linear pNOY102 plasmid DNAs (compare lanes 1, 8 and 9 in Figure 4). When incubated for 1 h at 37°C in the presence of fresh Na₂Se (15 µM), the plasmid was almost entirely converted into its nicked form.

**Figure 2. Analysis of cell-cycle phases distribution in asynchronous cultures of wild-type (wt) and rad52Δ strains.** Before analysis by flow cytometry, cells were grown for 2 h in YTD in the presence of the indicated concentrations of Cpt or sodium selenide. The histogram shows the number of cells (counts) corresponding to each fluorescence intensity (in arbitrary units). The percentages of G1 and G2/M subpopulations are indicated above the corresponding peaks. The boxes at the top of the panels indicate the fluorescence intensity intervals chosen to calculate G1 and G2/M cell populations.

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form (compare lanes 10 and 8 in Figure 4). In contrast, sodium selenite did not break DNA (lane 2) unless it was mixed with glutathione (lane 4), a condition known to convert selenite into hydrogen selenide [13,35]. Thus, we conclude that selenide is sufficient to break phosphodiester bonds, whereas selenite alone has no detectable effect.

Free Radical Production is Required for Selenide to Nick DNA

To determine whether ROS were involved in selenide-induced DNA breakage, the above experiment was repeated in the presence of a radical quencher or of detoxification enzymes. Before addition of sodium selenide or sodium selenite, the plasmid was mixed with either SOD (which converts superoxide anions $O_2^-$ to $H_2O_2$ and $O_2$), catalase (which converts $H_2O_2$ into $H_2O$ plus $O_2$) or mannitol (a quencher of various radicals including 'OH, but, importantly, not $O_2^-$. The electrophoretic profiles of plasmid DNA samples recorded after treatment with either sodium selenide or sodium selenite plus glutathione were not modified by the presence of catalase or SOD (compare lanes 6 and 7 to lane 4, and lanes 12 and 13 to lane 10 in Figure 4). On the other hand, in the presence of mannitol, the profiles remained similar to that observed when sodium selenide was omitted (compare lanes 4 and 11 to lane 8 in Figure 4). These data indicate that one or several free radicals, with the exception of $O_2^-$, are involved in the selenide-induced DNA damaging reaction.

ESR spectroscopy was used to monitor the production of radicals from sodium selenide. Reaction of the spin trap DEPMPO with 'OH radicals leads to an oxidized stable form (DEPMPO-OH) which has a characteristic 8-peak spectrum [36]. Superoxide anions also react with DEPMPO, but the resulting modified spin trap (DEPMPO-OOH) displays a clearly distinct ESR spectrum [36]. We first carried out a control ESR analysis after incubation of DEPMPO with $H_2O_2$ and Fe(II)-EDTA. This mixture is expected to lead to 'OH production through the Fenton reaction. Consistent with 'OH formation in this control experiment, the spectrum of DEPMPO-OH was observed (Figure 5A). In the presence of mannitol (150 mM) in the mixture, the magnitude of the spectrum was strongly reduced (Figure 5B). This confirmed that the spectrum in Figure 5A indeed reflected 'OH production.

**Figure 4. Effect of sodium selenide on DNA integrity in vitro.** Purified pNOY102 plasmid DNA was submitted to native gel electrophoresis after incubation during 1 h at 37°C with different combinations of the indicated compounds: 15 µM Na$_2$Se, 25 µM sodium selenite (Na$_2$SeO$_3$), 500 µM glutathione (GSH), 200 units/ml SOD, 80 mM mannitol, 50 units/ml catalase, 200 units/ml topoisomerase I (Topo I) or 500 units/ml Sall. The different topomers, as detected after ethidium bromide staining, are indicated on the left. The star on the right side of the figure points a contaminant systematically present in our pNOY102 preparations. The numbers at the bottom of the figure show the ratios between nicked and supercoiled DNA band intensities. With topoisomerase, the ratio is between relaxed and supercoiled DNA.
To analyze radical production in the presence of selenide, DEPMPO was incubated in the presence of Na$_2$Se (100 μM). The eight-peak spectrum characteristic of DEPMPO-OH was obtained (Figure 5C). Because DEPMPO-OOH can slowly convert to DEPMPO-OH [36], we performed the same experiment in the presence of SOD to make sure that the DEPMPO-OH spectrum did not arise from a conversion of DEPMPO-OOH to DEPMPO-OH. We also added catalase to the reaction mixture to preclude formation of hydroxyl radicals from SOD-produced H$_2$O$_2$. Indeed, parasitic generation of OH radicals might have occurred in the case of contamination of the solution by trace amounts of transition metals. Under these conditions (SOD and catalase), the eight-peak spectrum of DEPMPO-OH was again obtained (Figure 5D). Similarly to what we observed in the Fenton reaction control experiment, the amplitude of the spectrum strongly decreased when the mixture of DEPMPO and Na$_2$Se was supplemented with mannitol (Figure 5E). Thus, we conclude that the reaction of selenide with dioxygen produces free radicals, likely 'OH radicals, and that if superoxide ions were produced, their concentration remained below the detection threshold.

**DNA Damage by Sodium Selenide Requires the Presence of Dioxygen**

To establish whether the DNA-damaging reaction required the presence of dioxygen, we compared the rates of selenide-induced SSB formation under aerobic and anaerobic conditions, *in vitro*. In the experiment shown in Figure 5A, increasing amounts of Na$_2$Se were added to the pNOY102 plasmid DNA solution. Incubations were performed inside a glove box with dioxygen partial pressure continuously lower than 5 ppm. DNA solutions were prepared in the glove box in either a deoxygenated buffer (anaerobic conditions) or an oxygenated one (aerobic conditions). The reaction was initiated by mixing the DNA solutions with Na$_2$Se, and quenched after 1 min by addition of 150 mM mannitol.

In the oxygenated buffer, in agreement with the results presented in Figure 4, addition of increasing amounts of sodium selenide caused progressive nicking of supercoiled DNA, as shown by the decreasing intensity of the corresponding gel band (Figure 6, right side). The nicking of plasmid DNA was nearly complete in the presence of 0.1 mM Na$_2$Se. In contrast, supercoiled DNA remained unmodified by sodium selenide in deoxygenated buffer (Figure 6, left side). We conclude that selenide-induced DNA breakage strictly requires the presence of dioxygen.

**Dioxygen in the Culture Medium Potentiates the Toxicity of Sodium Selenide *in vivo***

Because selenide requires dioxygen to nick DNA *in vitro*, we asked whether dioxygen was necessary for selenide to cause death of yeast cells. The wild-type *S. cerevisiae* strain was exposed to various concentrations of Na$_2$Se (0–50 μM) for 5 min, under aerobic or anaerobic conditions. To estimate short-term viability, cells were plated on rich medium just after the treatment and their ability to form colonies was determined. As expected, in the presence of dioxygen, Na$_2$Se concentrations higher than 5 μM induced a significant loss of viability (Figure 7). When cells were maintained in strict anaerobic conditions, cell death was no longer observed in the presence of sodium selenide. This experiment establishes that selenide toxicity indeed implies an oxygen-dependent mechanism.

**Discussion**

**Free Radicals are Produced During Oxidation of Hydrogen Selenide by Dioxygen**

This study provides experimental evidence that, in the presence of dioxygen, hydrogen selenide (H$_2$Se/Se$_2^-$) damages DNA *in vitro*. Implication of free radicals in the damaging reaction is indicated by the protective effect afforded by mannitol. ESR analysis confirms the production of radicals from selenide.

Aqueous hydrogen selenide reacts with dissolved dioxygen, resulting in the formation of red colloidal selenium (Se$_8$ ring), according to the following overall reaction:

$$8 \text{HSe}^- + 4 \text{O}_2 \rightarrow \text{Se}_8 + 8 \text{OH}^-$$

This reaction, which is completed in a few minutes in aerobic conditions, obeys a very complex kinetics [37] indicative of a free...
radical chain mechanism. Putative intermediate species include superoxide ion, hydrogen peroxide and polyselenides [37].

The closely related reaction between aqueous hydrogen sulfide and dioxygen is also believed to be autocatalytic [38,39]. It is thought to start with the formation of sulfide radicals, followed by the production of polysulfide radicals which become active sites for oxygen chemisorption. Many additional reactions may take place, leading to the formation of radicals such as HSO3\(\cdot\), HSO4\(\cdot\), HSnSO3\(\cdot\) and maybe also \(\cdot\)OH.

Because of the chemical similarity between sulfur and selenium, several radicals, including selenium-containing radicals, are expected to be produced during oxidation of aqueous selenide by dioxygen. Thus, assignment of a particular radical to the breakage of DNA is difficult. ESR experiments are compatible with the formation of \(\cdot\)OH radicals upon selenide reaction with dioxygen. Inhibition by mannitol of the selenide-induced DNA breakage also suggests the production of \(\cdot\)OH. However, involvement in the DNA breaking reaction of some other undetermined radical cannot be formally excluded. In any case, our results exclude that superoxide anions account for DNA breakage. Indeed, mannitol, which does not quench superoxide anions, fully protects DNA in the nicking assay, and SOD addition has no effect in the same assay.

**Hydrogen Selenide Induces DSBs in vivo**

The in vitro experiments with plasmid DNA raise the question as to whether the presence of hydrogen selenide originating from sodium selenide causes a breaking of phosphodiester bonds in vivo. If DSBs are indeed produced, proteins involved in the cellular response to DSBs (repair machinery and checkpoint proteins) should be important for cell survival in the presence of sodium selenide. Actually, in our genome-wide screen, we observed that the absence of a single protein participating in HR (the main and ubiquitous pathway for DSB repair) frequently caused hypersensitivity to sodium selenide. Another evidence for DSB induction upon sodium selenide treatment comes from cell-cycle analysis by flow cytometry. In the cellular context, a single DSB is enough to activate the G2/M checkpoint [40]. We indeed observed that exposure to Na2Se blocked the cells in G2/M. The percentage of blocked cells increased with the Na2Se concentration in the culture medium. The effect was amplified if HR was impaired, as shown with a \(\text{rad}52\) deletion mutant. DSB production could finally be evidenced in vivo using PFGE analysis. We thus propose that the production of hydrogen selenide from Na2Se induces DSBs, either by producing nearby SSBs on opposite strands or through replication of an SSB-containing DNA. Such a conclusion is reminiscent of that obtained with selenite [21,22]. Because conversion of selenite to selenide is necessary to nick DNA in vitro, it is likely that selenite has to be reduced to selenide to exert its toxicity in vivo. This idea has already been developed elsewhere [9,13].

**DSBs are the Main Cause of Cell Death in the Presence of Sodium Selenide**

As mentioned above, different radicals are likely to be produced during the oxidation of selenide by O2. Therefore, several types of DNA damages may occur. Here, we obtain evidence that DNA fragmentation is the main cause of cell death. Firstly, sodium selenide addition is accompanied by a marked G2/M block. This
behavior is expected with DSBs, not with DNA damages such as base oxidations. Secondly, the spectrum of genes associated with hypersensitivity to sodium selenide, as identified in our screen, resembles the spectra obtained with γ-ray [30] or Cpt treatments (Figure S3) which, each, predominantly induce DSBs. Thirdly, this spectrum is clearly different from the spectra obtained with several other physical or chemical genotoxic agents [31,41]. Although we did not find genes of the BER pathway in the first 250 ranks, we cannot formally exclude an underestimation of the importance of this pathway in our approach, given the functional redundancies between proteins implicated in BER. Nevertheless, we believe that, in the case of the response to sodium selenide, the BER pathway is not as important as in the case of the response to other DNA-damaging agents such as UV. Indeed, a screen analysis of gene-disrupted yeast cells showed that disruption of APN1 (a key gene in the BER pathway) confers hypersensitivity to UV [41]. In our screen, this gene is found at the 306th rank.

Only a Few Genes of the Oxidative Stress Response Regulon are Involved in the Resistance to Sodium Selenide

Our data clarify the role of glutathione in selenium toxicity. A priori, glutathione may play two opposite roles in the selenide-induced oxidative stress [42]. On the one hand, glutathione is capable of scavenging various radicals including •OH, and can therefore help the cell to face a selenium challenge. It can also repair proteins modified in the course of the selenide redox cycling. On the other hand, reduced glutathione can regenerate hydrogen selenide from elemental selenium [43], a compound itself produced by the oxidation of selenide by O2. Such a cycle is likely to accentuate the toxicity of hydrogen selenide. In our screening experiment in the presence of Na2Se, the GSH1, GLR1, GRX1 and GRX3 genes, involved in glutathione and glutaredoxin homeostasis, occupy ranks 1, 3, 7 and 31, respectively. Therefore, glutathione clearly emerges as an important player in the protection against sodium selenide. This suggests that the protective role of reduced glutathione predominates over its deleterious effect. Many other proteins belonging to the oxidative stress response, such as catalase, SOD, thioredoxins or glutathione peroxidases, are not determinant for the resistance to sodium selenide, since none of their genes ranks under 250 (Figure 1).

Our data pinpoint the importance of the DNA repair pathway in the response to sodium selenide exposure. Surprisingly, the genes involved in this pathway were not detected in a transcriptome analysis of selenite-stressed yeast cells [14]. Possibly, this absence reflects some differences between the cellular responses to selenide or selenite. Another explanation might be that, prior to a toxic challenge, corresponding gene products are already at sufficient concentrations in a cell to confer protection. Indeed, a study dealing with the identification of genes important for the survival of S. cerevisiae exposed to well known DNA-damaging agents (ionizing radiation, UV radiation, cisplatin, H2O2) concluded that transcription of most of the genes involved in protection against DNA damage were not stimulated in response to toxic doses of the damaging agent [44]. This may explain why the DNA repair pathway has been overlooked in gene expression profiling experiments.

Supporting Information

Figure S1 Strategy for quantitative analysis of a cell population using a two-step amplification of DNA barcodes. Barcode regions are in gray. In the first PCR, one oligonucleotide hybridizes to a sequence beyond the barcode region (U1 for the upstream barcode, D1 for the downstream one) and the other one hybridizes inside the kanMX cassette (KU and KD, respectively). The second amplification is performed in the presence of fluorescent primers U2/D2 annealing inside the first amplicons, and in the presence of unlabeled U1/D1 primers. The PCR products (and thus the corresponding barcodes) can then quantified by microarray hybridization.

Figure S2 Fitness defect analysis. (A) Distribution of the relative fitness defect values (log2(s)) obtained with 4320 mutants of the systematic deletion collection. The observed distribution of fitness defect values (bins of size 0.01) was fitted to a Cauchy-Lorentz function by non-linear least squares regression. Part of the histogram and the fitted values are shown in the inset to illustrate the asymmetry of the results distribution, with an imbalance towards negative values (corresponding to strains that grew poorly in the presence of Na2Se). (B) Comparison of observed fitness defects obtained on pairs of strains that are deleted at the same locus. Data include 294 pairs of mutants for which there is an overlap of at least one nucleotide between the two corresponding deleted ORFs (non-parametric Kendall test, p<2.2·10−16).

Figure S3 Correlation between the relative fitness defects associated with sodium selenide and Cpt treatments. The data of the present study were compared with the fitness scores associated with Cpt treatment [1]. The comparison involves 4386 genes. The gray area corresponds to genes whose invalidation leads to fitness parameters smaller than −0.5 in the cases of the two toxic compounds. This comparison indicates a good correlation between the behaviors of the mutants in response to the two toxic agents (non-parametric Kendall test, p = 3.2·10−14). 1. Hillemeyer ME, Fung E, Wildenhain J, Pierce SE, Hoon S, et al. (2008) The chemical genomic portrait of yeast: uncovering a phenotype for all genes. Science 320:362–365.

Table S1 Selenide sensitivity data on the systematic haploid deletion collection of S. cerevisiae strains.

Table S2 Weights used to filter the microarray data.

Methods S1

Methods S2

Acknowledgments

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Author Contributions

Conceived and designed the experiments: GP CS MD ML AJ SB PP. Performed the experiments: GP MD FB ML LD PP. Analyzed the data: GP CS MD XL CM SB PP. Wrote the paper: GP CS SB PP.
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