Defects in purine nucleotide metabolism lead to substantial incorporation of xanthine and hypoxanthine into DNA and RNA.
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Deamination of nucleobases in DNA and RNA results in the formation of xanthine (X), hypoxanthine (H), oxanone, and uracil, all of which are miscoding and mutagenic in DNA and can interfere with RNA editing and function. Among many forms of nucleic acid damage, deamination arises from several unrelated mechanisms, including hydrolysis, nitrosoative chemistry, and deaminase enzymes. Here we present a fourth mechanism contributing to the burden of nucleobase deamination: incorporation of hypoxanthine and xanthine into DNA and RNA caused by defects in purine nucleotide metabolism. Using Escherichia coli and Saccharomyces cerevisiae with defined mutations in purine metabolism in conjunction with analytical methods for quantifying deaminated nucleobases in DNA and RNA, we observed large increases (up to 600-fold) in hypoxanthine in both DNA and RNA in cells unable to convert IMP to XMP or AMP (IMP dehydrogenase, guaB; adenylosuccinate synthetase, purA, and ADE12), and unable to remove dITP/ITP and dXTP/XTP from the nucleotide pool (dITP/XTP pyrophosphohydrolase, rdgB and HAM1). Conversely, modest changes in xanthine levels were observed in RNA (but not DNA) from E. coli lacking purA and rdgB and the enzyme converting XMP to GMP (GMP synthetase, guaA). These observations suggest that disturbances in purine metabolism caused by known genetic polymorphisms could increase the burden of mutagenic deaminated nucleobases in DNA and interfere with gene expression and RNA function, a situation possibly exacerbated by the nitrosative stress of concurrent inflammation. The results also suggest a mechanistic basis for the pathophysiology of human inborn errors of purine nucleotide metabolism.

The chemical modification of nucleobases in DNA and RNA can arise from both physiological and adventitious mechanisms at all stages of nucleic acid metabolism. This is particularly true for deaminated versions of the nucleobases. As shown in Fig. 1 for purines, nucleobase deamination in DNA and RNA leads to the formation of 2′-deoxy- and ribonucleoside forms of hypoxanthine (2′-deoxyinosine, dI; inosine, Ino) from adenine, xanthine (2′-deoxoxanthosine, dX; xanthisine, Xiao) and oxanone (2′-deoxoxanone, dO; oxanone, Oxo) from guanine, and uracil (2′-deoxyuridine, dU; uridine, Urd) from cytosine (1). All of these products are miscoding and mutagenic in DNA (2–4) and can interfere with RNA editing (5) and the function of noncoding RNAs (6). There are three recognized mechanisms that contribute to nucleobase deamination in DNA and RNA, the simplest of which is hydrolysis (7). A second source of nucleobase deamination is associated with the nitrosative stress caused by increases in nitric oxide-derived nitrous anhydride during inflammation (1). A third mechanism is associated with deaminase enzymes acting on RNA and DNA, with activation-induced cytidine deaminase converting cytidine to uridine during immunoglobulin diversification in B lymphocytes and adenosine deaminases responsible for mRNA editing (5) and modification of tRNA and rRNA (8). Here we propose a fourth mechanism in which perturbation of purine nucleotide metabolism leads to incorporation of the purine intermediates hypoxanthine and xanthine into DNA and RNA.

Purine nucleotide metabolism plays a central role in cell physiology of both prokaryotes and eukaryotes (e.g., ref. 9). While purine biosynthesis contributes to many facets of cell metabolism, the present work focuses on synthesis of the purine nucleotide components of RNA and DNA, which is highly conserved in all organisms (10). As shown in Fig. 2 for Escherichia coli, purine anabolism is composed of a 10-step reaction starting with phosphoribosylpyrophosphate (PRPP) and ending with inosine monophosphate (IMP). Conversion of IMP to AMP is mediated by adenylosuccinate synthetase (purA) and followed by adenylosuccinate lyase (purB). GMP is also derived from IMP but via xanthosine monophosphate (XMP) through the action of IMP dehydrogenase (guaB) and GMP synthetase (guaA). Xanthine and hypoxanthine thus play significant roles in purine metabolism as components of the cellular nucleotide pool.

The concentrations of purine nucleotide pool components are highly regulated (11, 12), with many human diseases caused by imbalances arising from genetic polymorphisms and mutations in purine metabolism (13–17). For example, loss of adenosine deaminase results in severe combined immunodeficiency (18), while loss of hypoxanthine-guanine phosphoribosyltransferase leads to the hyperuricemia and neurological symptoms of Lesch–Nyhan syndrome (14), and increases in the activity of purine nucleotide metabolic enzymes, such as IMP dehydrogenase, are associated with several types of cancer (19). Further, genetic polymorphisms in purine metabolic enzymes are associated with the toxic side effects of thiopurine drugs (17). One possible mechanism underlying these pathologies and diseases involves the forma-
tion of noncanonical nucleotide triphosphates from nucleotide pool components, with subsequent incorporation into DNA and RNA. From the standpoint of mutagenesis, this would parallel the polymerase-induced misincorporation of canonical 2′-deoxyribonucleotides into DNA (e.g., ref. 20). Similar polymerase-induced misincorporation has been shown to occur with ribonucleotides during DNA synthesis (21), which leads to a potential mechanism for insertion of ribonucleotide forms of xanthine and hypoxanthine into DNA as well as RNA.

Given the potential for the nucleotide pool to affect genomic integrity, cells possess mechanisms to prevent incorporation of damaged nucleoside triphosphates into nucleic acids and to remove noncanonical nucleotides from DNA. For example, ribonucleotide insertions into DNA are repaired by a combination of RNase H and topoisomerase I in eukaryotic cells (22), while removal of xanthine and hypoxanthine from DNA is accomplished in E. coli by repair enzymes such as endonuclease V (EndoV; nfi), endonuclease VIII (EndoVIII; nei), alkyladenine glycosylase A (AlkA; alkA), and mismatch-specific uracil DNA glycosylase (MUG; mug) (23–26). Misinsertion of damaged nucleotides is partially controlled by polymerase substrate specificity, but a variety of enzymes have evolved to remove noncanonical nucleotide triphosphates from the nucleotide pool. E. coli possesses two nucleoside-triphosphatases, YjjX and RdgB, that cleave (d)XTP and (d)ITP to diphosphate (YjjX) and monophosphate (RdgB) forms (27–29), which parallels E. coli MutT pyrophosphorylase activity that acts on 8-oxo-dGTP (30). RdgB homologs in S. cerevisiae, mice, and humans (ITPA) possess similar activities (29, 31). Indeed, loss of ITPA in mice, though perinatal lethal, leads to increased incorporation of hypoxanthine in DNA and RNA (32, 33), which is alleviated by expression of the IDP-hydrolyzing activity of nudix-type motif 16 protein (NUDT16) (34).

The hypothesis tested here is that, beyond the enzymes that cleanse the nucleotide pool, defects in the biosynthetic network of purine metabolic enzymes lead to increased incorporation of xanthine and hypoxanthine into DNA and RNA. To test this model, we used chromatography-coupled isothe dilution tandem mass spectrometry to quantify misincorporation of the deaminated nucleobases into the DNA and RNA of E. coli and S. cerevisiae strains possessing mutations in purine nucleotide metabolism. The results reveal that disruption of critical nodes in the purine metabolism network causes large increases of hypoxanthine, but not xanthine, in DNA and RNA. These results have implications for the pathophysiological mechanisms underlying many human metabolic disorders and suggest that disturbances in purine metabolism caused by known genetic polymorphisms could increase the burden of mutagenic deaminated nucleobases in DNA and interfere with gene expression and RNA function, a situation possibly exacerbated by the nitrosative stress of concurrent inflammation.

Results
Quantification of Deaminated Nucleotides in DNA and RNA. To complement our method for quantifying dX, dI, dO, and dU (Fig. 1) (35), we developed an isotope-dilution LC-MS/MS method for quantifying their ribonucleoside equivalents. This involves hydrolysis of RNA, HPLC purification of ribonucleosides (Fig. S1), and their quantification by LC-MS/MS using predetermined molecular transitions. While quantitative rigor is ensured with isotopically labeled internal standards, DNA and RNA deamination artifacts were minimized with deaminase inhibitors (35).

The results of analysis of xanthine and hypoxanthine in RNA and DNA in E. coli strains are shown in Table 1 and those for S. cerevisiae in Table 2. In all studies, the G deamination products dO and Oxo were below detection limits (<5 oxanine per 10^6 nt), which is consistent with previous studies in human cells and mouse tissues (36, 37). It was also apparent that the levels of xanthine and hypoxanthine were higher in wild-type E. coli RNA than in DNA, by twofold and ninefold, respectively. This is not surprising because Ino is one of many ribonucleoside modifications in tRNA and rRNA (8). As a negative control for these studies, we observed that the level of dU in wild-type and mutant E. coli strains was constant at approximately 5 per 10^7 nt, which is consistent with mutations that do not involve pyrimidine metabolism.

Defects in Purine Nucleotide Metabolism Increase the Levels of Xanthine and Hypoxanthine in RNA and DNA. Focusing first on data for hypoxanthine in E. coli (Table 1), it is apparent that the loss of individual E. coli genes leads to substantial increases in the levels of dI and Ino. Loss of either enzyme that acts on IMP to initiate formation of guanine nucleotides (guaB) or adenine nucleotides (purA) causes twofold to sevenfold increases in Ino and smaller
and hypoxanthine nucleoside triphosphatase in the in vivo substrate specificities need to be reconsidered. Finally, both enzymes have in vitro activities against (d)XTP and (d)ITP. YjjX nucleoside triphosphatase, even in combination with loss of side triphosphates from the pool. However, loss of the similar cleansing enzymes suggests that generation of (d)ITP exceeds the enzymatic capacity increases are not prevented by the (d)ITP-hydrolyzing RdgB, which dI and Ino, but this is not the case (Table 1). Further, these in-...would suggest that loss of both IMP dehydrogenase (guaB) and adenylosuccinate synthetase (purA) would lead to higher levels of dl and Ino, but this is not the case (Table 1). Further, these increases are not prevented by the (d)ITP-hydrolyzing RdgB, which suggests that generation of (d)ITP exceeds the enzymatic capacity of the pool cleansing enzymes.

Not surprisingly, loss of the RdgB nucleoside triphosphatase led to a 10-fold and fivefold increase in Ino and dl, respectively, which is consistent with its role in removing deaminated nucleoside triphosphates from the pool. However, loss of the similar YjjX nucleoside triphosphatase, even in combination with loss of RdgB, had no effect on xanthine or hypoxanthine levels (Table 1). Both enzymes have in vitro activities against (d)XTP and (d)ITP (27–29), so the results suggest that RdgB is the major xanthine and hypoxanthine nucleoside triphosphatase in E. coli and that the in vivo substrate specificities need to be reconsidered. Finally, the loss of either the nfi gene encoding the EndoV DNA repair protein or the alkA gene encoding the AlkA DNA glycosylase did not affect the levels of xanthine or hypoxanthine in DNA or RNA; the implications of this observation will be discussed shortly in the context of DNA repair.

While the loss of single genes in purine metabolism led to substantial changes in hypoxanthine levels, losses of specific combinations of genes led to synergistic increases in the content of hypoxanthine in DNA and RNA. The most striking effect occurred with loss of both rdgB and purA genes, which led to large increases in dl and Ino ranging from 155- to 642-fold. This can be rationalized as a loss of PurA causing an increase in IMP concentra-

<table>
<thead>
<tr>
<th>Genotype</th>
<th>Ino per 10^6 nt</th>
<th>dl per 10^6 nt</th>
</tr>
</thead>
<tbody>
<tr>
<td>Wild type</td>
<td>11 ± 3</td>
<td>2.8 ± 0.5</td>
</tr>
<tr>
<td>purA</td>
<td>25 ± 4'</td>
<td>3.0 ± 0.8</td>
</tr>
<tr>
<td>guaB</td>
<td>74 ± 14'</td>
<td>1.5 ± 0.3'</td>
</tr>
<tr>
<td>rdgB</td>
<td>100 ± 20'</td>
<td>3.3 ± 0.6'</td>
</tr>
<tr>
<td>yjjX</td>
<td>13 ± 3</td>
<td>3.1 ± 0.4</td>
</tr>
<tr>
<td>nfi</td>
<td>11 ± 1</td>
<td>2.6 ± 0.2</td>
</tr>
<tr>
<td>purA/guaB</td>
<td>39 ± 24</td>
<td>1.2 ± 0.03'</td>
</tr>
<tr>
<td>purA/nfi</td>
<td>30 ± 5'</td>
<td>3.6 ± 0.8</td>
</tr>
<tr>
<td>purA/rdgB</td>
<td>2,100 ± 200'</td>
<td>2.8 ± 0.7</td>
</tr>
<tr>
<td>guaB/rdgB</td>
<td>813 ± 125'</td>
<td>1.5 ± 0.9'</td>
</tr>
<tr>
<td>rdgB/yjjX</td>
<td>80 ± 20'</td>
<td>3.3 ± 0.4</td>
</tr>
<tr>
<td>rdgB/nfi</td>
<td>100 ± 35'</td>
<td>3.5 ± 0.7</td>
</tr>
<tr>
<td>purA/aln</td>
<td>1,700 ± 300'</td>
<td>7.2 ± 0.5'</td>
</tr>
<tr>
<td>purA/rdgB</td>
<td>2,900 ± 400'</td>
<td>2.1 ± 2.1</td>
</tr>
<tr>
<td>purA/guaB</td>
<td>30 ± 7'</td>
<td>1.1 ± 0.5'</td>
</tr>
<tr>
<td>purA/rdgB</td>
<td>2,100 ± 200'</td>
<td>2.2 ± 0.6</td>
</tr>
<tr>
<td>purA/rdgB/nfi</td>
<td>1,900 ± 400'</td>
<td>6.5 ± 0.3'</td>
</tr>
<tr>
<td>purA/rdgB</td>
<td>240 ± 400'</td>
<td>3.3 ± 0.4</td>
</tr>
<tr>
<td>purA/rdgB</td>
<td>1,971 ± 212'</td>
<td>1.2 ± 0.06'</td>
</tr>
</tbody>
</table>

*Data represent mean ± SD for n = 3; units of lesions per 10^6 nt. 
†Significantly different from wild type at P < 0.05 by Student's t test.

Table 2. Levels of Ino and dl in genomic DNA and total RNA from S. cerevisiae lacking purine nucleotide metabolism genes*

<table>
<thead>
<tr>
<th>Genotype</th>
<th>Ino per 10^6 nt</th>
<th>dl per 10^6 nt</th>
</tr>
</thead>
<tbody>
<tr>
<td>Wild type</td>
<td>42 ± 8</td>
<td>2.0 ± 0.1</td>
</tr>
<tr>
<td>ade12</td>
<td>35 ± 5</td>
<td>3.0 ± 0.6</td>
</tr>
<tr>
<td>ham1</td>
<td>40 ± 10</td>
<td>2.2 ± 0.2</td>
</tr>
<tr>
<td>ade12/ham1</td>
<td>1,500 ± 72'</td>
<td>98 ± 20'</td>
</tr>
</tbody>
</table>

*Data represent mean ± SD for n = 3. 
†Significantly different from wild type at P < 0.05 by Student's t test.
human rdgB homolog, ITPA. A common polymorphism in ITPA, the 94C > A allele present in 11–19% of Asians, reduces enzyme activity by 50% (16) and is associated with toxic responses to thiopurine therapy for cancer, autoimmune disease and inflammatory bowel disease (17), presumably due to increased incorporation of thiopurines into DNA and RNA. This parallels the fivefold and 10-fold increases in hypoxanthine in DNA and RNA, respectively, caused by loss of rdgB in E. coli.

Compared to the loss of purA, guaB or rdgB as individual genes, disruption of a combination of early and late enzymes produced synergistic increases in hypoxanthine incorporation into DNA and RNA (Table 1). Specifically, combined loss of rdgB and either purA or guaB, which individually produced twofold to ninefold increases in hypoxanthine in RNA, caused 80- to 190-fold increases (Table 1). A similar synergy was observed in yeast with loss of ADE12 and HAM1 (Table 2). These synergies suggest that even modest changes in the activities of pairs of these enzymes caused by genetic polymorphisms could result in increased misincorporation of hypoxanthine into nucleic acids in humans. Given the large reductions in activity caused by common genetic polymorphisms in the human homologs of purA and rdgB (15, 16), a substantial number of humans could possess polymorphisms that affect two or more purine metabolism genes, with the consequence of increased incorporation of deaminated nucleotides into DNA and RNA.

An intriguing feature of these studies was the lack of effect of metabolic defects on nucleic acid levels of xanthine, which could be due to any of several possible mechanisms. For DNA, it is possible that dI and dX are repaired with different efficiencies by the various activities of EndoV, EndoVIII, AlkA, and MUG (23–26). However, while DNA repair efficiency could account for some portion of the differing levels of xanthine and hypoxanthine in DNA, their differing levels in RNA suggest other mechanisms. Three examples support a model in which xanthine- and hypoxanthine-containing nucleotides interact differently with purine metabolic enzymes. First, xanthine nucleotides may bind less effectively in allosteric feedback mechanisms that are known to be important for regulation of ribonucleotide reductases (39). As another example, since the purA/guaA/rdgB mutation should cause reduced levels of ATP and GTP, reductions in ATP-dependent NAD+ synthesis could lead to decreased activity of the NAD+-dependent IMP dehydrogenase (guaB), which would reduce XMP synthesis. A third possibility is that xanthine-containing nucleotides are poorer substrates for kinases than hypoxanthine species. Agarwal et al. showed that GMP, dGMP, S-azaGMP, and IMP are substrates for human guanylate kinase while XMP, UDP, CMP, and 6-thioIMP are not (40, 41). Thus, lower levels of (d)XTP may be due to less efficient phosphorylation of XMP.

Two other mechanisms may account for reduced xanthine incorporation into nucleic acids in the event that the levels of nucleoside triphosphates are equivalent. First, it is possible that (d)XTP are poorer substrates than (d)ITP for RNA and DNA polymerses, respectively, which is supported by the known selectivity of several polymerses (42, 43). Second, there may be a cryptic xanthine nucleoside triphosphatase beyond RdgB and YjjX. These alternatives await quantification of the components of the nucleoside triphosphate pool.

The phenomenon of purine metabolic defects leading to incorporation of deaminated nucleotides into nucleic acids should have pathological consequences in a variety of cellular processes. With respect to mutagenesis, while there is some controversy about the ability of hypoxanthine to induce mutations in E. coli (4, 44), dI, dX, and dU are all established mutagens in higher organisms. So it is reasonable to assume that genetic polymorphisms that raise the steady-state levels of dX and dI in DNA in humans would exacerbate the toxicity associated with the nitrosative stress of chronic inflammation, with further increases in dX and dI exceeding the cell’s repair capacity and contributing to mutations and cancer risk.

Considering both coding and noncoding RNA species, the incorporation of xanthine and hypoxanthine into RNA should have pathological consequences for gene expression. For example, editing of mRNA by ADAR-mediated conversion of adenine to hypoxanthine is a highly regulated mechanism controlling gene expression (5). The insertion of Ino into mRNA could thus be interpreted as inappropriate editing, with consequential sequestration of the mal-edited mRNA into heterochromatin (45) or entry into RNAi pathways (5). Given the role of Ino as an abundant posttranscriptional modification of RNA, misinsertion of Ino into tRNA and rRNA could affect translational fidelity and efficiency, as supported by recent observations (e.g., refs. 46 and 47).

Finally, increased concentrations of (d)XTP and (d)ITP could lead to enhanced incorporation of ribonucleotides into DNA and 2′-deoxyribonucleotides into RNA. The former has been observed with canonical ribonucleotides as a result of polymerase infidelity and leads to genomic instability if the levels of incorporated ribonucleotides exceed the capacity of RNase H and other repair enzymes to correct the insertion errors (21, 48). The combined effect of incorrect nucleotide class and noncanonical nucleobase caused by insertion of IPT or XTP into DNA could further complicate the pathophysiology of the polymerase errors.

Our observations suggest that disturbances in purine metabolism could increase the burden of mutagenic deaminated nucleotide bases in DNA and interfere with gene expression and RNA function, a situation possibly exacerbated by the nitrosative stress of concurrent inflammation. The results also suggest a mechanistic basis for the pathophysiology of human inborn errors of purine nucleotide metabolism.

Materials and Methods
Materials and Instruments. Chemicals and reagents were used without further purification unless otherwise noted and were obtained as follows: Nuclease P1: USBiological and Roche Diagnostic; coforornycin: National Cancer Institute; uniformly 15N-labeled Adu, Guo, DA, dG and DC: Cambridge Isotope Laboratories (Andover, MA); phosphodiesterase I: USB; alkaline phosphatase, dexferoxamine, RNase A, ammonium acetate, tetrahydroydouridine, DNase I: Sigma Chemical; UV-grade acetonitrile: Honeywell Burdick & Jackson; Milli-Q system for all water purification and Microcon YM-10 filters: Millipore; Genomic-tip 100/G kits, RNeasy Mini kits and RNAlater reagent: Qiagen (Valencia, CA); Instruments used: Agilent Bioanalyzer series 2100, Agilent 1100 HPLC system (LC), Agilent LC/QQQ 6460 triple quadrupole mass spectrometer (MS/MS), Applied Biosys API 3000 MS/MS and Agilent LC/QTOF 6520 quadrupole time-of-flight mass spectrometer (QTOF).

Synthesis of Isotopically Labeled Internal Standards. Syntheses of [15N2]-labeled dX, dI, and dD are described elsewhere (35). Xao, Ino, Oxo were synthesized similarly starting with uniformly 15N-labeled Adu and Guo. All standards were purified by HPLC with a Phenomenex LUNA C18 column (250 mm × 4.6 mm, 5 μm, 100 Å pore), characterized by QTOF mass spectrometry, and quantified by UV absorbance (35, 49).

Preparation of E. coli and S. cerevisiae Mutants. E. coli mutants were constructed in a W3110 background by transduction using bacteriophage P1 vir. Individual mutant genes were obtained from the Keio Collection (50), as gene knockouts carrying a kanamycin gene cartridge, and were transduced into E. coli W3110 using selection for kanamycin resistance, with the resistance cartridge removed using flp sequences surrounding the kanamycin cartridge and flp recombinase (51). Additional rounds of transduction and gene cartridge removal yielded the multiple mutant strains. The phenotypes of purA mutants and guaA and guaB mutants were confirmed by an inability to grow in the absence of Ade and Gua, respectively. The alkA mutant phenotype was confirmed by sensitivity to methyImethane sulfonate and genotypes of rdgB, yggX, and mfl mutants were confirmed by PCR.

The S. cerevisiae strains are from the Saccharomyces Genome Deletion Project (52). To construct the ham1::URA3 ade12::KANMX4 double mutant strain, a two-step cloning process was performed. In the first step, a ham1::KANMX4 DNA sequence was amplified from S. cerevisiae ham1 mutant genomic DNA using the 5′ primer GCGCGCGGAA TTCCTTTTTCAGCATGAAATCGGC-
TA and the 3′-primer GGGCGCGGATCCTGTAGCTGCAGTCAGTAACATTC. PCR amplified products were digested with EcoR1 and ligated into EcoR1-cleaved pUC19. The two internal ClaI restriction sites in the KANMX4 gene were utilized to construct the second clone, for which the URA2 gene was amplified from CYP225 tapm1 URA3 genomic DNA using the 5′-primer CTGCGCCAAATCCTGTAGCTGCAGTCAGTAACATTC. PCR products were digested with ClaI and ligated into ClaI-cleaved ham1::KANMX4 pUC19 vector, with the ham1::URA3 DNA sequence subsequently amplified using the same primers for the ham1::KANMX4 sequence in step one. To obtain the ham1::URA3 ade12::KANMX4 double mutant, the amplified sequence was transformed into a S. cerevisiae ade12::KANMX4 mutant strain (Geitz Lab Yeast Transformation Kit Quick and Easy protocol). Plating on yeast nitrogen base agar plates lacking uracil was used to select for ham1::URA3 ade12::KANMX4 transformants. The phenotype of ade12 mutants was confirmed by their inability to grow in the absence of Ade and that of ham1 mutants by their sensitivity to 6-hydroxylaminopurine.

DNA and RNA Purification. Following collection of bacterial and yeast, genomic DNA was purified (Genomic-tip 100 G kit) in the presence of deaminase inhibitors cofomycin (0.5 μg/mL) and tetrahydrodouridine (125 μg/mL) to avoid nucleobase deamination artifacts (35). Total RNA was isolated from bacteria (Qiagen RNeasy Mini Kit) with cofomycin and tetrahydrodouridine. DNA and RNA were initially quantified by UV absorbance for preparation of hydrolysates and subsequently, for normalization of mass spectrometry data, by quantification of canonical nucleosides during HPLC resolution using external calibration curves.

DNA and RNA Hydrolysis. Total RNA or genomic DNA (50 μg) was lyophilized and redissolved in 30 μL of ammonium acetate buffer (30 mM, pH 6.2) to which zinc chloride (10 μM, 10 mM) and [15N]-labeled internal standards (10 pmol) were added to yield a final volume of 50 μL. DNA was hydrolyzed by addition of nuclease P1 (4 U) and DNAseI (5 U) with incubation at 37 °C for 3 h, followed by dephosphorylation by addition of ammonium acetate buffer (30 mM, pH 7.4), cofomycin (0.5 μg/mL), tetrahydrodouridine (125 μg/mL), alka-line phosphatase (15 U) and phosphodiesterase I (1.0 U) and incubation at 37 °C for 6 h. The enzymes were subsequently removed by Microcon YM-10 filtration. Total RNA was processed similarly, with the exception of initial hydrolysis in 30 mM ammonium acetate and 2 mM ZnCl₂ (pH 6.8) with nuclease P1 (1 U) and RNAse A (5 U) for 3 h at 37 °C and dephosphorylation with alkaline phosphatase (10 U) and phosphodiesterase I (0.5 U) for 1 h at 37 °C following addition of acetate buffer to 30 mM, pH 7.8.

Preparation of X- and 1-Containing Nucleosides. dl, dX, dO, and dU in DNA were resolved by reversed-phase HPLC as noted earlier at a flow rate of 0.4 mL/min with a stepwise acetonitrile gradient in 4 mM ammonium acetate (pH 7.4): 0–40 min, 1–8% acetonitrile; 40–45 min, 8–50%; 45–50 min, 50–70% (reversal); 45–51 min, 70–100%; 51–61 min, 100%. Individual nucleosides were collected at predefined retention times: dl, 19 min; dX, 27 min; dO, 34 min; and dU, 45 min; for canonical nucleosides: dC, 22 min, dG, 36 min, T, 39.5 min, and dA, 50.5 min. Ino and Xao in RNA hydrolysates were also purified by HPLC using the same Phenomenex Synergi C18 column (250 × 4.6 mm, 5 μm particle, 80 Å pore), with a flow rate of 0.5 mL/min and acetonitrile gradient in 8 mM ammonium acetate (pH 6.9) at 36 °C: 0–18 min, 1–2%; 18–23 min, 2%; 23–28 min, 2–7%; 28–30 min, 7%; 30–32 min, 7–100%; 31–41 min, 100%. Individual nucleosides were collected at experimentally determined retention times (min): Xao, 22; Ino, 31; Oxo, 40; canonical nucleosides eluted as follows (min): Cyd, 16; Urd, 17.5; Guo, 32; 157 and 141 for internal standards [15N]N-1-X/O and I; at m/z 269, 253, and 229 for dX/dO, dl, and dU, respectively. The product ions were monitored in Q3 at m/z 157, 141, and 115 for internal standards [15N]N-1-X/O-I, and U; at m/z 153, 137, and 111 for Xo/I, and U, respectively. Linear calibration curves were obtained daily.

Ribonucleosides were injected onto the same HPLC column eluted isocratically at 200 μL/min with an aqueous mobile phase consisting of 0.1% acetic acid and 1% acetonitrile. The eluent was analyzed by Agilent 6410 operated with an electrospray ion source at 350 °C and in positive ion mode, with wide resolution for Q1 and unit resolution for Q3, and other optimized parameters: ion source voltage, 4.0 kV; nebulizer gas, 8; curtain gas, 8; collision gas (N₂), 4; declustering potential, 20; focusing potential, 100; entrance potential, 5; collision energy, 10; collision cell exit potential, 10. MMM was used for detection of nucleosides with a dwell time to 200 ms. Q1 was set to transmit the precursor ions MH⁺ at m/z 273, 257, and 231 for the internal standards [15N]N-1-X/O, dl, and dU, respectively: at m/z 269, 253, and 229 for dX/dO, dl, and dU, respectively. The product ions were monitored in Q3 at m/z 157 and 141 for internal standards [15N]N-1-X/O-I and at m/z 153 and 137 for Xo/I and U, respectively. The limits of detection for Ino, Xao, and Oxo were 5, 10, and 50 fmol, respectively.

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