The genome-wide transcriptional response to varying RpoS levels in *Escherichia coli* K-12.

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Running title: Effect of RpoS concentration on *E. coli* transcriptome

Abstract

The alternative sigma factor RpoS is a central regulator of a many stress responses in *Escherichia coli*. The level of functional RpoS differs depending on the stress. The effect of these differing concentrations of RpoS on global transcriptional responses remains unclear. We investigated the effect of RpoS concentration on the transcriptome during stationary phase in rich media. We show that 23% of genes in the *E. coli* genome are regulated by RpoS level, and we identify many RpoS-transcribed genes and promoters. We observe three distinct classes of response to RpoS by genes in the regulon: genes whose expression changes linearly with increasing RpoS level, genes whose expression changes dramatically with the production of only a little RpoS ("sensitive" genes), and genes whose expression changes very little with the production of a little RpoS ("insensitive"). We show that sequences outside the core promoter region determine whether a RpoS-regulated gene in sensitive or insensitive. Moreover, we show that sensitive and insensitive genes are enriched for specific functional classes, and that the sensitivity of a gene to RpoS corresponds to the timing of induction as cells enter stationary phase. Thus, promoter sensitivity to RpoS is a mechanism to coordinate specific cellular processes with growth phase, and may also contribute to the diversity of stress responses directed by RpoS.

Importance

The sigma factor RpoS is a global regulator that controls the response to many stresses in *Escherichia coli*. Different stresses result in different levels of RpoS production, but the consequences of this variation are unknown. We describe how changing the level of

RpoS does not influence all RpoS-regulated genes equally. The cause of this variation is likely the action of transcription factors that bind the promoters of the genes. We show that the sensitivity of a gene to RpoS levels explains the timing of expression as cells enter stationary phase, and that genes with different RpoS sensitivities are enriched for specific functional groups. Thus, promoter sensitivity to RpoS is a mechanism to coordinate specific cellular processes in response to stresses.

Introduction

Genome-wide measurements of RNA levels have revolutionized our understanding of how cells organize their patterns of transcription. These studies have given us snapshots of how patterns of gene expression change in response to changes in the external environment. They have also allowed us to define the regulons controlled by specific transcription factors. A major weakness of the vast majority of these studies is that they explore the function of a regulatory protein only by comparing expression of target genes in a wild-type strain to a gene knock-out, or to a mutant with a single diminished or increased level of activity. While some genetic regulatory networks are certainly switch-like (1, 2) and can be fully characterized by only two levels of activity, many other regulatory proteins vary continuously in their abundance and/or activity. How regulons respond to a range of regulator levels is a largely unstudied question (3). Should we expect all genes in a regulon to increase or decrease expression by the same relative amount following a change in abundance of a regulatory protein? Or should we expect genes to respond in different ways? These questions motivate this study.

A paradigmatic example of a bacterial regulatory protein whose abundance and activity vary continuously in response to different conditions is the alternative sigma factor RpoS in *Escherichia coli*. Transcription by RNA polymerase containing RpoS is responsible for the general stress response (4–6). Under conditions of optimal growth in the laboratory (such as exponential phase growth in rich media at 37°C), RpoS levels are nearly undetectable in the model *E. coli* strain K-12. As conditions become poorer for growth, either because cells begin to starve for various nutrients, or because they face physical challenges such as low temperature or elevated osmolarity, RpoS levels rise (4–6). RpoS coordinates the transcription of genes that are critical for the response to these stresses.

RpoS expression is not an all-or-none phenomenon. For example, RpoS levels rise continuously as cells transition from exponential growth to stationary phase (7). Moreover, starvation for different nutrients upregulates RpoS to differing levels (8, 9). This level of control over RpoS levels is accomplished by regulating transcription, translation, and protein degradation (4, 7, 10), allowing for careful control over protein levels. In addition to regulation of protein abundance, RpoS activity can also be directly modulated by a number of factors, such as Crl (11).

Not only do RpoS levels vary across conditions for a single strain, but different strains of *E. coli* also differ in their patterns of expression of RpoS. For example, naturally occurring strains can differ in the amount of RpoS they produce during exponential phase (12) or stationary phase (13). All studies that have measured RpoS levels in naturally occurring strains of *E. coli* have detected variation (13–17), though

97 the extent and cause of this variation in RpoS between strains is still a matter of some
98 controversy (16, 17).

99 Microarray studies (18–20) have shown that RpoS controls the expression of at least
100 500 genes (over 10% of the genome) either directly or indirectly, but the set of RpoS-
101 regulated genes differs across environmental conditions. For most RpoS-controlled
102 genes, it is not known whether the gene is directly transcribed by RpoS or regulated
103 indirectly, as a consequence of RpoS transcribing other genes. Previous studies have
104 not investigated the impact of changes in RpoS levels on the RpoS regulon, or whether
105 quantitative differences in RpoS levels between environmental conditions influence the
106 observed differences in what genes are RpoS-regulated. It is clear that *E. coli* has a
107 complicated regulatory network to fine-tune RpoS levels to different conditions, but we
108 do not yet know the consequences of this regulation.

109 In this study, we tested the hypothesis that members of the RpoS regulon vary in
110 their response to RpoS levels. Using a combination of ChIP-seq and RNA-seq, we
111 identified RpoS-regulated genes, and we showed that genes vary in their sensitivity to
112 RpoS levels in a manner dependent on sequences outside of the core promoter region.
113 Sensitivity of genes to RpoS levels corresponds to the order in which genes are induced
114 during the transition into stationary phase, and genes with different levels of sensitivity
115 are enriched for specific functional groups. Thus, the level of sensitivity of genes to
116 RpoS controls the physiological response to different stress conditions.

117

118 **Materials and Methods**
119

120 *Culture conditions*

121 Cells were grown in 5mL of LB broth (1% tryptone, 0.5% yeast extract, 1% NaCl) in
122 150 x 18 mm tubes, positioned vertically and shaking at 225 rpm at 37°C in a water bath,
123 unless otherwise specified in the text. When required, antibiotics were used at the
124 following final concentrations: ampicillin at 100 µg/ml (for plasmids) or 25 µg/ml (for
125 chromosomal integration); chloramphenicol at 20 µg/ml; kanamycin at 50 µg/ml.
126
127 *Strains and genetic manipulations*
128 Strains and plasmids used in this study are listed in Table S1. The wild-type genetic
129 background for this all experiments except for ChIP-seq is BW27786, a strain designed
130 to give a graded transcriptional response to increasing arabinose concentration (21). To
131 create a strain of this background lacking *rpoS*, the Δ*rpoS746::kan* allele of JW5437
132 (22) was moved by P1 transduction into BW27786, creating strain DMS2545.
133 The arabinose-inducible RpoS strain was created by PCR amplifying the *kan* gene
134 and the P_{araB} promoter of plasmid pAH150 (23) using primers ParaBRpoSRecomb-F
135 and ParaBRpoSRecomb-R (Table S2). This PCR product was then integrated into the
136 *nlpD* gene (i.e. 5' of *rpoS*) in a MG1655 background using plasmid pKD46 (24), and P1
137 transduced into BW27786, creating strain DMS2564. This strain thus lacks both the
138 native transcriptional and translational control of RpoS.
139 For flow cytometry experiments, plasmid pDMS123 (25), which contains a *otsB-gfp*
140 transcriptional fusion, was transformed into DMS2564 by the method of Groth et al. (26).
141 ChIP-seq experiments used strain RPB104, an unmarked derivative of MG1655 that
142 expresses a C-terminally SPA-tagged derivative of RpoS from its native locus. This

143 strain was constructed by P1 transduction of kan^R-linked *rpoS*-SPA from a previously
144 described strain (27). The kan^R cassette was removed using the pCP20 plasmid that
145 encodes Flp recombinase (24).
146
147 *Construction and chromosomal integration of lacZ fusions*
148 The *gadB* and *astC* plasmids were built by standard cloning methods. The *gadC*
149 and *astA* promoter regions with transcription factor binding sites were PCR-amplified
150 with primers gadCpromoter +/- and astApromoter +/- (Table S2), which included *Kpn*I
151 and *Eco*RI restriction sites for cloning. Cloning of core promoter regions was performed
152 by annealing oligonucleotides designed to contain the whole RpoS binding region, as
153 predicted by Fraley *et al.* (28) and Castanie-Cornet and Foster (29). Oligonucleotides
154 were annealed by heating 1 µM of forward and reverse primer for one minute at 100°C
155 with 5 mM $MgCl_2$ and 7 mM Tris-HCl and then cooling slowly to room temperature.
156 Inserts and plasmid (pLFX) were digested with *Eco*RI-HF and *Kpn*I-HF (NEB), ligated
157 with T7 ligase (NEB) and cloned into strain BW23473. Transformants were miniprepped
158 and inserts were verified by Sanger sequencing.
159 The *gadA* and *hdeA* plasmids were built using Gibson assembly (30) with the
160 NEBuilder HiFi Assembly kit (New England Biolabs). The *gadA* and *hdeA* promoter
161 regions were PCR amplified with primers hdeAHiFi+/- and gadAHiFi+/-. The core
162 promoter was cloned with the single long oligo hdeAcoreHiFi or gadAcoreHiFi, as
163 predicted by Arnqvist et al (31) and De Biase et al (32). PCR products or
164 oligonucleotides were mixed with pLFX digested with *Kpn*I-HF and *Eco*RI-HF, and
165 assembled according to the manufacturers' instructions. Mixtures were cloned into

166 strain BW23473, transformants were miniprepped, and inserts were verified by Sanger
167 sequencing.
168 *lacZ* fusion plasmids were integrated into strain DMS2564 with helper plasmid
169 pPFINT (33). Single-copy integrants were confirmed using the PCR assay of Haldimann
170 and Wanner (23).
171
172 *Quantitative Western Blotting*
173 Quantitative western blotting was used to measure RpoS levels. Cells were
174 inoculated from frozen cultures into 5 mL of LB, and grown overnight at 37 °C, shaken
175 at 225 rpm. 5 µL of this overnight culture was diluted into 5 mL of LB with the
176 appropriate concentration of arabinose, and grown for 20 h. 100 µL of overnight culture
177 to be assayed was centrifuged and resuspended in 1X Laemmli sample buffer (Sigma-
178 Aldrich) and boiled for 5 minutes. Samples were diluted 1:10 in 1X Laemlli, and 10 µL
179 was electrophoresed on a 10% polyacrylamide gel (Bio-Rad) in tris-glycine running
180 buffer (25 mM Tris base, 250 mM glycine, 0.5% SDS) at 100 V for 90 min at room
181 temperature. Proteins were transferred to an Immobilon-FL PVDF membrane by
182 electrophoresis at 100 V for 45 min at 4 °C in transfer buffer (48 mM Tris base, 39 mM
183 glycine, 20% methanol, and 0.0375% SDS). Membranes were blocked by overnight
184 incubation in Odyssey blocking buffer (Li-Cor) at 4 °C.
185 The blocked membrane was probed with affinity-purified monoclonal antibodies
186 to RpoS (clone 1RS1) and RpoD (clone 2G10) (NeoClone) at a final concentration of
187 0.4 µg / mL in Odyssey blocking buffer plus 0.2% Tween 20 for 1 hour at room
188 temperature. The membrane was washed four times for five minutes each with 15 mL of

1X TBST. A fluorescent secondary antibody (IRDye 800CW goat anti-mouse, Li-Cor) was diluted 1:10,000 in a solution of Odyssey Blocking Buffer plus 0.2% Tween 20 and 0.01% SDS and incubated in the dark for 1 hour at room temperature. The membrane was washed as before, dried for 2 hours between sheets of Whatmann 3MM blotting paper, and imaged on a LiCor Clx fluorescent imager.

Band intensity was estimated using Image Studio 2.1 (LiCor). RpoS levels were divided by RpoD levels to normalize for differences in total protein levels. The ratio of RpoS to RpoD is biologically meaningful because this ratio, rather than RpoS level alone, dictates levels of transcription from RpoS-dependent promoters due to sigma factor competition (34).

RNA-seq experiments and analysis

Cells were inoculated from frozen cultures into 5 mL of LB, and grown overnight. 5 µL of this overnight culture was diluted into 5 mL of LB and grown for 20 hours. (For the intermediate 26% RpoS condition, a final concentration of 10^{-4}% arabinose was also added.) RNA was purified from 200 µL of overnight culture by pelleting and resuspending in 500 µL of Trizol at 65 °C, followed by purification on a column (Direct-Zol, Zymo Research). Samples received two 30-minute DNase treatments using TURBO DNA-free (Ambion) following the manufacturer's instructions. RNA samples were then purified on a column (RNA Clean & Concentrator, Zymo Research). Samples were stored at -80 °C until used. Three samples were prepped from each culture and pooled to generate sufficient RNA. Two biological replicates were prepared for each strain or condition of interest. rRNA depletion, cDNA synthesis, library preparation, and

sequencing were performed by a commercial provider (Otogenetics, Norcross, GA). Paired-end, 100 bp sequences were generated for 7-15 million reads per sample.

Before reads were mapped, the first ten base pairs of each read were trimmed using FASTX-Toolkit 0.0.13. Reads were mapped to the NCBI K12 reference genome (NC_000913.2 Escherichia coli str. K-12 substr. MG1655) using BWA v0.7.5a (35).The number of read pairs mapped to each gene was counted with HTSeq 0.6.1 (36). Differential expression analysis was performed with DESeq v2.13 (37). All p-values were first FDR adjusted using the procedure of Benjamini and Hochberg (38). p-values were then further Bonferroni-adjusted for the three comparisons between pairs of RpoS levels. All differential expression p-values reported in this paper reflect both the FDR and Bonferroni-adjustments.

To determine if a gene differed significantly from the null expectation of linearity, we calculated the probability of the observed read count value at 26% RpoS if the true expression level was given by the linear prediction between the endpoints. Note that to calculate both the expected read count and the probability of the observed value under the null (i.e. the p-value) our model required estimating both the DEseq size factor (for scaling) and the dispersion (a variance factor) for each of the samples. The negative binomial probability model (routinely used to measure count data, (e.g. (37)) was used, with the size factors and dispersion estimated from DESeq (37) to calculate the probability of the observed read count at 26% RpoS.

GO term analysis was performed using the topGO package (39) together with the org.EcK12.eg.db annotation package (40) in R 3.1.0. Enrichment was assessed using the weight01 algorithm (39) together with the Fisher's-exact test. The GO hierarchy

was pruned to include only nodes with at least five associated genes, as significance tests can be unstable for GO terms with fewer genes (39).

A Venn diagram of the number of significant genes in each condition was prepared with EulerAPE 3.0.0 (41).

Analysis of transcription factor binding site enrichment used data from RegulonDB (42). We divided RpoS regulated transcriptional units in RegulonDB into sets: sensitive, insensitive and linear. Then, for each transcription factor in the database we determined how many of the transcription units it regulates fall into each set. We compared this with the number of regulated transcription units within the whole RpoS regulon. A case of enrichment is where a transcription factor regulates a disproportionately large number of units in a particular set (e.g. the sensitive set) compared with what we would expect based on the RpoS regulon as a whole. We identified such cases using a one-tailed hypergeometric test. All p-values were then FDR adjusted using the procedure of Benjamini and Hochberg (38). Transcription factors that with an FDR-adjusted p-value < 0.05 that regulated at least two transcription units in the gene set were considered enriched.

ChIP-seq analysis

E. coli strain RPB104 (MG1655 with C-terminally SPA-tagged *rpoS*) was grown overnight in M9 minimal medium with 0.4% glycerol at 30 °C and then subcultured 1:100 in the same medium, and grown for 60 hours to saturation (OD_{600} of ~3). ChIP-seq using the M2 monoclonal anti-FLAG antibody was performed as described

257 previously (43). Regions of enrichment ("peaks") were identified as described previously
258 (44). Relative enrichment was reported as a "Fold Above Threshold" (FAT) score.
259 We used MEME-ChIP (Version 4.11.2; default parameters) to analyze enriched
260 regions identified by ChIP-seq (45) Regions within 100 bp were merged. The reported
261 sequence motif was identified by MEME (Version 4.11.2) (46), run within the MEME-
262 ChIP environment.
263 We used MEME (46) within the MEME-ChIP environment (Version 4.11.2; default
264 parameters except that only the given strand was analyzed) to identify enriched
265 sequence motifs in regions surrounding TSSs associated with RpoS ChIP-seq peaks.
266 We analyzed sequences from -45 to +5 relative to each TSS.
267 We identified directly RpoS-transcribed genes by requiring that (i) the gene start is
268 within 300 bp of a ChIP-seq peak, (ii) the gene is positively regulated by RpoS, as
269 determined by RNA-seq, (iii) no other positively regulated gene starts within 300 bp of
270 the ChIP-seq peak, (iv) there is no associated sequence motif or TSS (identified in (47))
271 that would be consistent with transcription in the opposite orientation. For regulated
272 genes, we determined whether other genes in the same operon were also regulated,
273 using a published operon list (48) for *E. coli*. Peaks were associated with transcription
274 start sites from a previous study (47) by identifying transcription start sites within 20 bp
275 of a peak.
276
277 *QPCR*
278 RNA was isolated as for RNA-seq, except that samples underwent three 30-minute
279 DNase treatments using TURBO DNA-free (Ambion). cDNA was made from the RNA

280 samples using SuperScript VILO master mix (Invitrogen) and stored at -20 °C for use in
281 quantitative real-type PCR (qPCR) reactions.
282 qPCR was performed using Power SYBR Green master mix (Invitrogen), 2 µL of
283 cDNA, and primers at 300 nM. Cycling was performed at 95 °C for 10 min; and 40
284 cycles of 95 °C for 15 sec, 58 °C for 15 sec, and 68 °C for 30 sec. Three control genes
285 (*ftsZ, pgm,* and *hemL*) were used in addition genes of interest. These genes were
286 selected using the approach of Vandesompele et al. (49). Details of this selection
287 process, including the other seven genes tested, are available in Supplementary Text 1,
288 Table S3, and Figure S1.
289 For each gene, a standard curve was made using the following amounts of genomic
290 DNA: 200 ng, 20 ng, 2 ng, 200 pg, 20 pg, and 2 pg. Genomic DNA was extracted from
291 overnight cultures using a Puregene Kit (Qiagen), following the manufacturer's
292 instructions. Expression levels for each gene were interpolated from the standard curve.
293 Expression levels of experimental genes were divided by the geometric mean of the
294 three control genes.
295 Statistical assessment of sensitivity was performed by a bootstrapping approach.
296 Bootstrapping, rather than a parametric approach, was appropriate because the data
297 did not conform to parametric assumptions. In this approach, the unit of resampling was
298 the RNA isolated from the 0%, 26% and 100% RpoS conditions on an individual day.
299 For each resampled data set, the median expression at 0% and 100% RpoS was
300 calculated. The linear fit from the 0% RpoS median and the 100% RpoS median was
301 then used to predict the level of expression at the intermediate level of RpoS. Repeating
302 this re-sampling of the data 10,000 times yielded a 95% confidence interval for 26%

303 RpoS. The observed median for 26% was then compared to the confidence interval to
304 test for significance.
305
306 *Beta-galactosidase activity*
307 Beta-galactosidase activity was measured using the method of Miller (50). With
308 lacZ fusion strains, the level of sensitivity was quantified, rather than only categorized
309 as sensitive, linear, or insensitive. To quantify the level of sensitivity, for each replicate
310 we calculated the distance between the observed expression at the intermediate RpoS
311 concentration and the expected level based on a linear pattern, standardized by the
312 difference in expression between high and low RpoS conditions. Testing if sensitivities
313 were different from zero used a one-sample t-test, with p-values adjusted for multiple
314 comparisons using Holm's sequential adjustment method (51). Testing if two
315 sensitivities were different used a two-sample t-test with the same method of adjustment
316 for multiple comparisons.
317
318 *Analysis of published RNA-seq data*
319 We analyzed published RNA-seq data for wild-type and Δ*rpoS E. coli* over a
320 time-course of growth into stationary phase (48). Using normalized genome coverage
321 information extracted from wiggle files, we calculated relative abundance for all genes at
322 each of the four stationary phase time-points in the growth curves (the last four time-
323 points for each strain). We arbitrarily selected a threshold coverage value of 500 and
324 excluded any genes scoring below this threshold at all four time-points in wild-type cells.
325 This reduced variability associated with low expression levels. We excluded any genes

for which coverage at the final time-point was 0 since this would have prevented normalization. We also excluded any gene for which the first stationary phase time-point had the highest expression value of the four time-points for wild-type cells, since RpoS-dependent expression of these genes is likely to be masked by other factors. We then selected genes whose expression we had found to be induced by RpoS, and separated these genes into insensitive, linear, and sensitive classes. We calculated expression levels for each of these genes relative to expression at the final time-point.

Accession numbers

RNA-seq data were deposited in GEO with accession number GSE87856. ChIP-seq data were deposited in ArrayExpress with accession number E-MTAB-5339.

Results

The RpoS regulon in late stationary phase.

To understand the role of the RpoS protein in late stationary phase, we used RNA-seq to compare the transcriptome of wild-type and Δ*rpoS* cells. We observed differential expression of 1044 genes (23% of genes) between these two conditions ($p < 0.05$). Of the 1044 genes whose expression is influenced by RpoS, 605 are upregulated, and 439 are downregulated (Table S4).

Influencing transcription of 23% of the genome could have many potential phenotypic effects. To better understand the function of these genes, we examined which kinds of gene functions, as described by the gene ontology (GO), are more

abundant in the regulon than expected by chance. GO enrichment analysis indicates that the RpoS regulon includes many genes involved in metabolic processes (Table 1); 17 of 18 significantly enriched GO terms are metabolic terms. This metabolic reorganization includes the upregulation of genes encoding glycolytic enzymes and pathways for metabolism of L-arginine to glutamine, or from L-arginine to putrescine and then into succinate. RpoS also drives the downregulation of genes involved in the TCA cycle. These patterns of metabolic regulation are very similar to those identified in late stationary phase in *Salmonella enterica* (52). The only significant GO term not explicitly linked to metabolism (GO:0006970, “response to osmotic stress”) also includes metabolic genes, such as *otsA* and *otsB,* that are involved in trehalose biosynthesis.

Central metabolism is not the only phenotype similarly regulated by RpoS in both *S. enterica* and *E. coli*. Other similarities include transcription of genes involved in antioxidant activities, iron regulation, and Fe-S cluster assembly, the upregulation of proteases, and down regulation of porins. As in *S. enterica,* RpoS in *E. coli* influences the expression of many genes encoding other regulatory proteins, including *csrA*, *arcA, cra, fur, ihfA, hupA* and *hupB.* These proteins regulate phenotypes including carbon storage (CsrA), central carbon metabolism (ArcA and Cra), iron homeostasis (FUR), and also play a central role in structuring the nucleoid (IHF, HU). Not all of the regulation is identical between *E. coli* and *S. enterica,* however. For example, Lévi-Meyrueis *et al.* (52) noted that RpoS appears to direct switching between many pairs of isozymes, where one isozyme is expressed under conditions when RpoS is abundant, and its partner isozyme is expressed when RpoS levels are low. While some pairs show

372 a similar pattern in *E. coli* (such as *tktA/B* and *acnA/B*), others (such as *fumA/fumC*) do
373 not show this pattern. The reason why these enzymes involved in central carbon
374 metabolism might show this pattern is not clear.
375
376 *Genome-wide binding profile of RpoS*
377 While RNA-seq identifies which genes are regulated by RpoS, it cannot distinguish
378 between direct and indirect effects. To determine sites where RpoS binds (and hence
379 likely plays a direct role in transcription), we used ChIP-seq to map the association of
380 RpoS across the *E. coli* chromosome during stationary phase growth in minimal
381 medium. To facilitate ChIP, RpoS was C-terminally SPA-tagged at its native locus. We
382 reasoned that RpoS would only be identified with promoter regions since it is likely
383 released from elongating RNAP complexes. We identified 284 peaks of RpoS ChIP-seq
384 signal covering 260 genomic regions (peaks within 100 bp of each other were merged).
385 217 of the RpoS-bound regions are intergenic, and 67 are located within genes. We
386 reasoned that annotated genes that are transcribed by RpoS would be positioned close
387 to an RpoS-bound region. Consistent with this, 213 RpoS-bound regions are ≤300 bp
388 upstream of an annotated gene start. These 213 regions include 27 that are intragenic.
389 In 79 cases, we observed an RpoS-bound region ≤300 bp from the starts of two
390 divergently transcribed genes.
391 We used MEME to search for enriched sequence motifs within the RpoS-bound
392 regions. We detected a highly enriched motif in 107 regions (Figure 1A); this motif
393 closely resembles the known -10 hexamer recognized by RpoS (CTAYACT, where the
394 central YA are not as conserved as the other positions (53)). Moreover, occurrences of

the motif are positionally enriched with respect to the ChIP-seq peak center (Figure 1B), indicating that the data have high spatial resolution. Note that the motifs tend to be located just upstream of the peak centers (Figure 1B), as we have observed previously for the *E. coli* flagellar Sigma factor, FliA (44). This presumably reflects the fact that the footprint of initiating RNA polymerase associated with RpoS is not centered on the -10 hexamer.

The identification of RpoS binding sites allowed us to better understand the role of RpoS in both positive and negative regulation. While similar proportions of the RpoS regulon are positively and negatively regulated by RpoS, it is not clear if this is true at the level of direct regulation. Only 19 genes within 300 bp of a binding site are negatively regulated by RpoS. 19 of the 286 such genes is *fewer* than we would expect by chance if binding sites were randomly distributed around the genome, given than 439 of 4513 genes in the genome are negatively regulated. On the other hand, 111 of 286 genes within 300 bp are positively regulated, a highly significant effect (Fisher's exact test, $p < 10^{-16}$). Thus, the binding profile of RpoS is consistent with direct positive regulation of many genes, but provides no evidence of direct negative regulation.

Identification of RpoS-transcribed genes and promoters

We combined the ChIP-seq and RNA-seq data to identify genes that are directly transcribed by RNA polymerase containing RpoS. Thus, we identified 123 RpoS-transcribed genes in 99 transcripts (Table S5), and compared them to other published analyses (18–20, 42, 47, 54–56). In some cases, we identified RpoS-bound regions upstream of genes that were not detected as being RpoS-regulated by RNA-seq. These

418 genes may have promoters that bind transcriptionally inactive RNA polymerase (57, 58).
419 (Inactive polymerase could be bound but physically blocked from transcription
420 elongation by the presence of a bound repressor (59), or could be poised, waiting for a
421 signal to transition to transcription elongation (60, 61).) Alternatively, the disparity
422 between RpoS binding and regulation could be explained by differences in growth
423 conditions between the ChIP-seq and RNA-seq experiments, or by the possibility that
424 the C-terminal SPA tag affects the function of the C-terminus of the protein in the
425 response to transcription activators (57, 61).

426 The high spatial resolution of sigma factor ChIP-seq can facilitate the
427 identification of specific promoters (44, 62) when combined with nucleotide resolution
428 transcription start site (TSS) maps. Using published TSS data from *E. coli* under
429 stationary phase conditions similar to those used for the ChIP-seq experiment (47), we
430 determined all pairwise distances between RpoS ChIP-seq peaks and stationary phase
431 TSSs. We observed a strong enrichment for peak-TSS distances ≤20 bp (Figure S2).
432 We infer that these 112 TSSs are RpoS-transcribed. Consistent with this, the putative
433 RpoS-transcribed TSSs are associated with -10 hexamers that have features expected
434 of RpoS promoters (Figure 1C). In some cases, RpoS promoters were identified ≤300
435 bp upstream of genes that were not RpoS-regulated in the RNA-seq data set. We
436 presume that these are RpoS-transcribed genes that otherwise escaped detection
437 because of differences in the growth conditions used for ChIP-seq and RNA-seq.
438
439 *The transcriptome at three different RpoS levels*

This first view of the RpoS regulon considers RpoS as either present or absent. RpoS levels vary continuously across environmental stresses (7), so we sought to better understand how the level of RpoS in the cell influences transcription of target genes. To do this, we placed the *rpoS* gene under the control of the arabinose-inducible promoter P_{araB}. This promoter was integrated just upstream of the native *rpoS* gene, placing transcription under the control of arabinose concentration and removing the 5' region that regulates translation of the native mRNA (4).

To measure the resulting arabinose-induced expression of RpoS, we employed quantitative western blotting. RpoS levels increased with increasing arabinose concentration, from undetectable RpoS levels, to levels similar to those in wild-type cells (Fig 2a). To confirm that expression was graded and not all-or-none in this system (21), we used flow cytometry to measure expression in individual cells. We transformed the arabinose-inducible RpoS strain with the plasmid pDMS123 (25), which contains the RpoS-dependent *otsBA* promoter fused to *gfp*. As expected, *gfp* expression increased with increasing arabinose concentrations, and at each expression level the population was unimodal (Fig 2b).

We measured the transcriptome in cells with 26% of wild-type levels of RpoS, achieved with the addition of 10^{-4} % arabinose to cells with our arabinose-inducible *rpoS* strain. Of the genes that are differentially expressed between 26% RpoS and either 0% (ΔrpoS) or 100% (wild-type), 95% are also differentially expressed between 0% and 100% (Fig 3) ($p < 0.05$).

Nearly all genes that are significantly differentially expressed have monotonically increasing or decreasing patterns of expression between three levels of RpoS. Only

two genes (*ytfR* and *ytfT*) have an expression level at 26% RpoS that is significantly higher than the expression at both 100% and 0% RpoS. The only two genes with expression lower in the 26% RpoS condition than either 100% or 0% are *nlpD* and *pcm*. These genes lie immediately upstream of *rpoS. nlpD* was removed from the genome during the construction the arabinose inducible RpoS strain. The *pcm* gene is still present, but the level of transcription was lowered by the genetic modification.

Classifying quantitative responses to RpoS levels

To explore how genes respond to changing levels of RpoS, we developed a new metric, *sensitivity.* Our null expectation was that gene expression would increase linearly with increasing RpoS concentration. We observed many genes in our RNA-seq data set (such as *osmY*) whose expression at intermediate RpoS levels falls on, or close, to a line drawn between the 0% and 100% RpoS conditions (Fig 4a). We refer to these genes as *linear* in their response to increasing RpoS levels. Other genes (such as *astA*) are transcribed more at 26% than would be expected based on their expression levels at 0% and 100% (Fig 4b). We refer to these genes as *sensitive*, because only a small amount of RpoS results in relatively high levels of transcription. In contrast, some genes (like *gadC*) are expressed at intermediate RpoS levels less than expected based on expression at 0% and 100% RpoS; such genes are referred to as *insensitive* (Fig 4c).

We identified 910 linear, 102 sensitive, and 32 insensitive genes. 96% of sensitive genes and 88% of insensitive genes are positively regulated by RpoS. In contrast, only 53% of linear genes are positively regulated by RpoS, a significant difference (chi-square test, $p < 0.001$).

To determine whether sensitive or insensitive genes are associated with specific physiological responses to increasing RpoS levels, we again used GO enrichment. We tested the null hypothesis that the functions of these genes are a random sample from the entire RpoS regulon (not the whole genome). The GO terms significantly enriched in the sensitive class are response to osmotic stress, cellular amino acid catabolic process, and fatty acid oxidation (Table 2). Several genes encoding regulators are among the sensitive genes, including *arcA*, which encodes a global regulator of respiratory metabolism, and *rssB,* which encodes the adaptor protein required for degradation of RpoS by ClpXP.

GO enrichment is less useful for understanding the possible function of the insensitive gene set. Three GO terms are enriched (Table 3), but only a few genes with these annotations are present in the insensitive gene set, and their enrichment probably reflects the relatively small number of insensitive genes. More strikingly, the insensitive genes include nearly all of the genes required for acid resistance system 2: the structural genes *gadA, gadB,* and, *gadC*, and the regulator of this system *gadE* (63, 64). In addition, the genes *yhiM, yhiD*, *hdeA, hdeB , hdeD, mdtE, mdtF,* all of which have been described as having roles in acid resistance (63, 64), are insensitive.

We used reverse transcription coupled to qPCR to confirm the expression patterns of two insensitive genes (*gadC* and *gadE*), and three sensitive genes (*prpR, prpD,* and *astA*). All genes were positively regulated by RpoS (Fig 5), and the median expression at 26% RpoS is consistent with RNA-seq expression patterns for all genes. We used a bootstrapping approach to assess if expression at 26% was significantly above or below the linear expectation at 26% RpoS. The expression of *gadC* was significantly

509 insensitive ($p < 10^{-4}$), and *prpR* expression was significantly sensitive ($p < 10^{-4}$). *astA*
510 expression was marginally significantly sensitive (p = 0.06 for sensitivity), while *gadE*
511 and *prpD* were not significantly different from the linear expectation (p = 0.10 and p =
512 0.56 respectively).
513
514 *Control of sensitivity of expression*
515 What makes one promoter sensitive to RpoS levels and another promoter
516 insensitive to RpoS levels? We hypothesized three possible mechanisms. First,
517 chromosomal location could determine the response to RpoS levels, as is known to
518 occur in the context of total transcription levels (65). Second, it is possible that the DNA
519 sequence of the core promoter drives the response. Finally, it is possible that the
520 binding of transcription factors upstream of the core promoter influences the response
521 to RpoS levels. To test these hypotheses, we cloned the promoters (including all
522 upstream transcription factor binding sites annotated in EcoCyc (66)) of four operons
523 into the *lacZ* fusion plasmid pLFX (33). The four promoters were the sensitive
524 *astCADBE*, and the insensitive *gadA, gadBC,* and *hdeAB-yhiD.* Plasmid pLFX
525 recombines into the lambda-attachment site, placing the fusion in a novel genomic
526 context. While we did not detect binding of RpoS upstream of *astC, gadA, gadB,* or
527 *hdeA* by ChIP-seq, this is likely due to the difference in growth conditions, since these
528 genes have been previously shown to be directly transcribed by RpoS (31, 67, 68).
529 The pattern of transcription of all four fusions was the same as observed for the
530 respective genes in the RNA-seq data (Fig 6 a-d). *astC* transcription was sensitive to
531 RpoS levels (one sample t-test, p = 0.04), while *gadA, gadB*, and *hdeA* transcription

were all insensitive (one sample t-test, $p = 2 \times 10^{-6}$, $p = 10^{-5}$, $p = 0.04$ respectively). Since all reporters were placed at the same genomic locus, this result suggests that genomic location is not the determinant of response to RpoS levels.

A second potential mechanism to explain the difference between sensitive and insensitive genes is interactions between RpoS and the core promoter sequence. For example, specific nucleotides (or combinations of nucleotides) might tend to confer sensitive or insensitive patterns of transcription. The majority of both sensitive and insensitive genes were not associated with RpoS-bound regions in the ChIP-seq experiment, suggesting that they are indirectly regulated by RpoS (Table 4). The fact that most sensitive and insensitive genes are not bound by RpoS argues against the hypothesis of direct RpoS-DNA interactions driving sensitivity. To see if specific sequence motifs were consistently associated with sensitivity, we used the discriminative motif search feature of DREME (69) to search for motifs that differed between sensitive and linear, or insensitive and linear regulatory sequences. There are 28 ChIP-seq peaks associated with an operon with at least one sensitive gene, and 4 ChIP-seq peaks associated with an operon with at least one insensitive gene. We found no motifs that distinguished these sets of sequences, although the small number of sequences would have restricted the power of such a test.

To directly test the hypothesis that the core promoter region of RpoS-transcribed genes is responsible for determining the sensitivity to RpoS levels, we cloned this short region from the *astC, gadA, gadB, hdeA* promoters into pLFX and recombined the plasmids into the chromosome. These core promoters had absolute levels of transcription much lower than the entire promoter that included upstream transcription

555 factor binding sites (Fig 6e - h). The core promoter sequence for *astC* alone was
556 somewhat less sensitive to RpoS levels than the full-length construct (two sample t-test,
557 $p = 0.08$), although not significantly so, probably due to the variability of expression from
558 the full-length *astC* reporter. The *gadA* and *gadB* core promoters differed significantly
559 from their full-length promoters (two sample t-test, $p= 0.01$ and $p < 10^{-6}$, respectively).
560 The *hdeA* core promoter (Fig 6h) is not RpoS-dependent, showing a decline in
561 expression of approximately 10% in the presence of RpoS. This is consistent with the
562 previous finding that the ability of RpoD (but not RpoS) to transcribe *hdeA* is repressed
563 by H-NS protein bound upstream of the promoter (70). The core promoter construct
564 lacks the native H-NS binding sites upstream, and so the selectivity is apparently lost.
565 Thus, the core promoters do not replicate the RpoS sensitivity of their whole promoter
566 sequences. In addition, the three RpoS-dependent core promoters (Fig 6 e - g) do not
567 differ from each other in sensitivity (ANOVA, $p = 0.32$). We conclude that the core
568 promoter is not responsible for RpoS sensitivity.

569 The *lacZ* fusions suggest that neither genomic location nor core promoter sequence
570 influence the sensitivity of a promoter. The remaining possible mechanism is the binding
571 of specific TFs. If this is the case, we might expect that sensitive and insensitive genes
572 are enriched for binding by different TFs. We looked for such enrichment and found that
573 the sensitive genes are enriched for binding by ArgR, Nac, and NtrC (FDR < 0.05). The
574 insensitive genes are enriched for binding by ArcA, FliZ, GadE, GadW, GadX, H-NS,
575 PhoP, RcsB, and TorR. This very large set of regulators occurs largely because these
576 proteins are all annotated as regulating some or all of the operons involved in AR2:
577 *gadA, gadBC, gadE, hdeAB-yhiD,* and *hdeD.* The action of GadE, GadW, and GadX

578 occurs primarily at these loci, while the other regulators have many additional known
579 binding sites that are not near the promoters of insensitive genes. This specific
580 enrichment for TFs highlights proteins that may be responsible for the sensitive or
581 insensitive patterns of expression.
582
583 *Sensitivity to RpoS determines the timing of induction during entry into stationary*
584 *phase*
585 We hypothesized that the degree of sensitivity to RpoS could impact the timing of
586 expression under conditions when levels of active RpoS increase, such as during entry
587 into stationary phase. A previous study used RNA-seq to monitor the transcriptome over
588 a time-course of growth, including four time-points in stationary phase (48). We
589 analyzed these data to determine whether insensitive, linear, and sensitive genes show
590 differences in the timing of induction. We selected only those RpoS-induced genes
591 whose transcription increased upon entry into stationary phase. (It total, there were 250
592 such linear genes, 90 sensitive, and 19 insensitive.) We then determined the pattern of
593 expression for each such gene over four time-points beginning at the onset of stationary
594 phase. Although there is considerable variability in the expression patterns of genes, as
595 a group the three classes show clear differences in the timing of induction (Figure 7a).
596 Specifically, sensitive genes are induced most rapidly, followed closely by linear genes.
597 In both cases, expression peaked early in stationary phase and fell between 30 and 180
598 minutes after entering stationary phase. In contrast, insensitive genes showed relatively
599 little change in expression until the final time-point, 180 minutes into stationary phase.
600 To determine the importance of RpoS on the patterns of gene expression, we repeated

601 the above analysis using data generated from a Δ*rpoS* strain. As expected, the large
602 difference in timing between the groups of genes was greatly diminished (Figure 7b).
603 We conclude that the sensitivity of a gene is associated with the timing of expression
604 during stationary phase in an RpoS-dependent manner.
605
606 **Discussion**
607
608 *Expanding the known set of RpoS-transcribed genes and promoters*
609 The *E. coli* RpoS regulon has been widely investigated using targeted and genome-
610 scale approaches. Most genome-scale studies have focused on genes whose
611 expression is altered in the absence of RpoS (18–20, 55, 56). Hence, these studies
612 cannot distinguish between genes that are transcribed by RpoS, and those that are
613 indirectly regulated. ChIP-seq affords a high resolution view of RpoS binding. By
614 combining ChIP-seq with RNA-seq, we have identified 123 RpoS-transcribed genes
615 with high confidence, considerably expanding the known RpoS regulon. Previous
616 studies have suggested a role for RpoS in direct repression of some target genes (47,
617 57, 71). While we observed negative regulation of 439 genes by RpoS, there were
618 fewer of these repressed genes associated with ChIP-seq peaks than expected by
619 chance, suggesting that direct negative regulation by RpoS is rare.
620 Only two previous studies have used ChIP methods to map RpoS binding genome-
621 wide in *E. coli*. The first used ChIP-chip to identify 868 RpoS-bound regions (47), many
622 more identified in our study but with considerably lower resolution (median peak length
623 of 324 bp for RpoS ChIP-chip). The second used ChIP-seq, but identified relatively few

RpoS-bound regions (54). Of the 63 RpoS-bound regions identified in that study, 41 are shared with those from our study.

The high resolution of ChIP-seq allowed us to identify specific promoter sequences recognized by RpoS. By combining ChIP-seq data with a TSS map, we identified many high-confidence RpoS promoters. These promoters are strongly enriched for the presence of a -10 hexamer, with sequence preferences consistent with several of the previously described features of RpoS promoters (53). Specifically, we observed a preference for a C at position -8 (within the -10 hexamer), a C at position -13 (immediately upstream of the -10 hexamer), and a TAA a positions -6 to -4 (immediately downstream of the -10 hexamer). Previous studies have suggested that RpoS promoters often contain a -35 hexamer (72), although the spacing relative to the -10 hexamer is considerably more variable than for σ^{70} promoters. However, we did not detect enrichment of a -35 hexamer-like sequence among RpoS promoters, suggesting that the requirement for this element is weak.

Sensitivity to RpoS affects timing of expression, and groups functionally related genes

As is true for many transcription factors, RpoS levels vary continuously across a wide range of conditions. Our data show that genes differ in the sensitivity of their response to RpoS levels. Moreover, whether a gene is sensitive or insensitive to RpoS levels is associated with its function, suggesting a physiological rationale for sensitivity. For example, insensitive genes include many of those involved in the glutamate-dependent acid resistance 2 system (AR2). These genes are of particular interest

because AR2 allows *E. coli* to survive a pH of 2, an important trait for its ability to pass through the stomach and colonize the gastrointestinal tract (73, 74). To some extent, the shared sensitivity of functionally related genes can be explained by operon structure, i.e. co-transcription of multiple functionally related genes from a single promoter. However, the phenomenon of shared sensitivity for functionally related genes extends beyond operons. For example, the insensitive genes involved in AR2 are transcribed from at least five different promoters (66).

The fact that functionally related genes often have similar patterns of sensitivity to RpoS suggests that sensitivity can serve as a mechanism to control the timing of gene expression, and hence to coordinate specific cellular processes as part of a response to environmental stresses. Consistent with this idea, we have shown that the sensitivity of genes to RpoS levels correlates with the timing of their expression. RpoS sensitivity may drive similar patterns of expression in response to other stresses. Different environmental stresses are known to upregulate RpoS to varying levels (8), suggesting that some insensitive genes may only be expressed under certain stresses. In addition to the effects on gene expression, sensitivity to RpoS may also impact the effects of mutations in *rpoS*, which have been seen to evolve in the lab (75, 76). We expect mutations attenuating RpoS to have the strongest effect on insensitive genes, and the weakest effect on sensitive genes.

Possible mechanisms of RpoS sensitivity

While the connection between RpoS sensitivity and the timing of gene expression is clear, the molecular basis of sensitivity is less so. Our data indicate that the genomic

location of these operons does not determine the expression pattern. Moreover, several lines of evidence suggest that direct interactions between RpoS and the core promoter are also not responsible for determining sensitivity. First, analysis of the ChIP-seq data for the sensitive and insensitive genes finds no motif that distinguishes between them. Second, core promoters from both sensitive and insensitive genes do not replicate the pattern of expression of the full-length promoters. Third, the core promoters of a sensitive operon (*astC*) and two insensitive operons (*gadA* and *gadB*) have indistinguishable patterns of sensitivity, suggesting that what was excluded from those constructs (i.e., binding sites of regulatory proteins) determines the shape of the relationship.

Given our finding that core promoter sequences cannot explain the difference in sensitivity between promoters, we suggest that sensitivity is largely due to the action of specific regulatory proteins bound upstream. If this hypothesis is correct, it could also explain the physiological coherence of these groups. For example, many insensitive genes are involved in the AR2 phenotype and are also regulated by GadX, GadW, and GadE (77–79). If one or more of these three regulators was directly responsible for the insensitive pattern of expression, then this could help to explain the physiological coherence of the insensitive group. The sensitive genes, being a larger group, have no obvious single regulator, although relatively little is known about regulators that function in stationary phase.

It is also possible that the physical properties of promoters play a role in this process, either alone or in concert with transcription factor binding. For example, the supercoiling state of promoters influences levels of transcription (80, 81) and has been implicated in

693 regulating RpoS-dependent transcription (82, 83). It is possible that differential response
694 of promoters to supercoiling levels, either due to interactions directly with RpoS or by
695 changes in the ability of transcription factors to bind (84), plays a role in determining the
696 sensitivity of a promoter. If this type of regulation plays a role, it must be due the
697 structure of the whole promoter itself, rather than supercoiling differences conferred by
698 genomic location (85). We know this because the full promoters cloned into *lacZ* fusions
699 are able to replicate both sensitive and insensitive patterns of transcription, even when
700 moved to the same chromosomal location..
701 RpoS responds to a wide variety of environmental cues, and regulates genes
702 responsible for many different kinds of responses. This work has demonstrated that one
703 facet of that response, the level of RpoS produced, has varying effects across the
704 entirety of the regulon. The level of RpoS produced in a stress response, together with
705 the action of other transcription factors, may help to tune the RpoS-dependent stress
706 response in ways appropriate for individual stresses.
707

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714

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970 **Table 1** Biological processes enriched in the RpoS regulon.
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GO ID	GO Term	Genes in genome	Observed in regulon	Expected in regulon	p-value
GO:0009063	cellular amino acid catabolic process	69	38	18.5	3.5e-07
GO:0006099	tricarboxylic acid cycle	20	15	5.35	8.3e-06
GO:0006096	glycolytic process	18	14	4.81	8.3e-06
GO:0006094	gluconeogenesis	13	9	3.48	0.0016
GO:0009441	glycolate metabolic process	6	5	1.6	0.0063
GO:0006090	pyruvate metabolic process	21	17	5.61	0.0180
GO:0006970	response to osmotic stress	28	13	7.49	0.0190
GO:0006542	glutamine biosynthetic process	10	6	2.67	0.0270
GO:0009060	aerobic respiration	65	33	17.38	0.0285
GO:0006006	glucose metabolic process	36	20	9.62	0.0317
GO:0016052	carbohydrate catabolic process	276	94	73.79	0.0326
GO:0009101	glycoprotein biosynthetic process	13	7	3.48	0.0342
GO:0019395	fatty acid oxidation	30	13	8.02	0.0354
GO:0000162	tryptophan biosynthetic process	11	6	2.94	0.0463
GO:0009246	enterobacterial common antigen biosynthesis	11	6	2.94	0.0463
GO:0042398	cellular modified amino acid biosynthesis	17	7	4.55	0.0472
GO:0009239	enterobactin biosynthetic process	6	4	1.6	0.0472
GO:0000270	peptidoglycan metabolic process	51	14	13.64	0.0475

973 **Table 2** Biological processes enriched in the sensitive genes.
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GO ID	GO Term	Genes in genome	Observed in sensitive set	Expected in sensitive set	p-value
GO:0006970	response to osmotic stress	13	4	0.79	0.0055
GO:0009063	cellular amino acid catabolic process	38	6	2.31	0.0210
GO:0019395	fatty acid oxidation	13	3	0.79	0.0385

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Table 3 Biological processes enriched in the insensitive genes.

GO ID	GO Term	Genes in genome	Observed in insensitive set	Expected in insensitive set	p-value
GO:0006412	translation	21	3	0.6	0.018
GO:0006164	purine nucleotide biosynthetic process	10	2	0.28	0.030
GO:0006805	xenobiotic metabolic process	13	2	0.37	0.044

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992 **Table 4** Contingency table of the sensitivity of a gene and if is promoter is or is not
993 bound by RpoS.
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	Sensitive	Linear	Insensitive
Bound by RpoS	31	87	5
Not bound by RpoS	71	823	27

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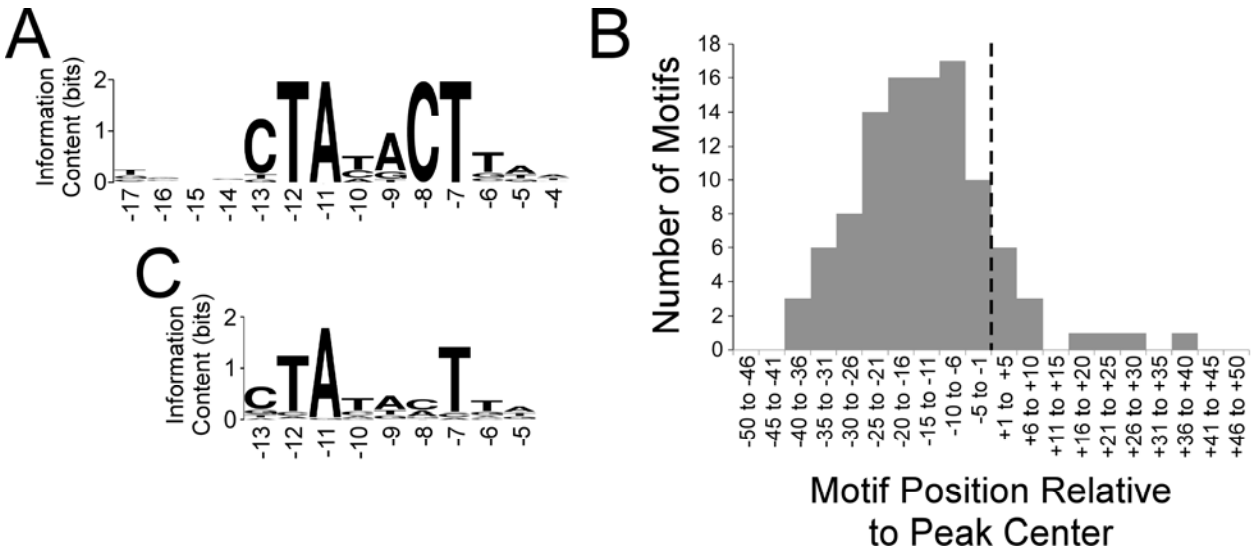

Figure 1. Analysis of RpoS ChIP-seq data. (A) Sequence logo (86) of the enriched sequence motif associated with RpoS-bound regions (MEME E-value 2 x 10^{-40}). The sequence logo was generated using WebLogo (87). Numbers below the motif refer to positions relative to the transcription start, based on known sequence preferences for RpoS (53). (B) Histogram showing the frequency distribution of distances between identified motifs and ChIP-seq peak centers. The black, vertical, dashed line indicates the peak center position. (C) Sequence logo (86) of the enriched sequence motif associated with TSSs located within 20 bp of RpoS ChIP-seq peak centers (MEME E-value 3 x 10^{-56}). Note that only a subset of the motif identified by MEME is shown, but positions outside the shown region all have information content below 0.2. The sequence logo was generated using WebLogo (87). Numbers below the motif refer to positions relative to the transcription start, based on known sequence preferences for RpoS (53).

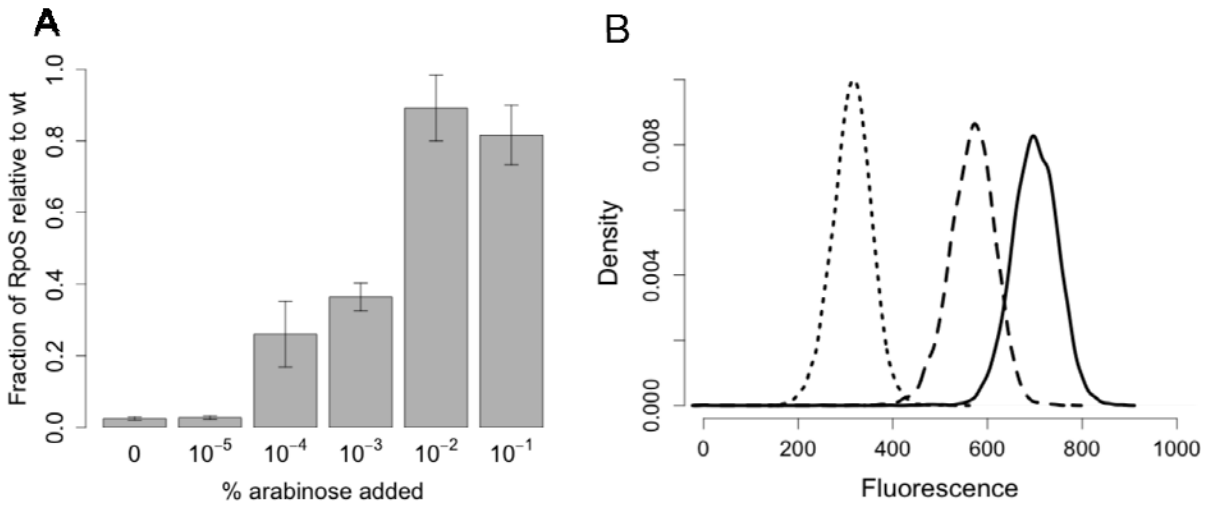

Figure 2. Arabinose control of RpoS protein levels in strain DMS2564. (a) Increasing arabinose results in more RpoS expression in strain DMS2564, as measured by western blotting. n = 4 to 5; error bars represent SEM. (b) Flow cytometric measurement of an *otsB-gfp* fusion under 0%, 10^{-4}%, and 10^{-1}% arabinose. (Dotted line, dashed line, and solid line, respectively.)

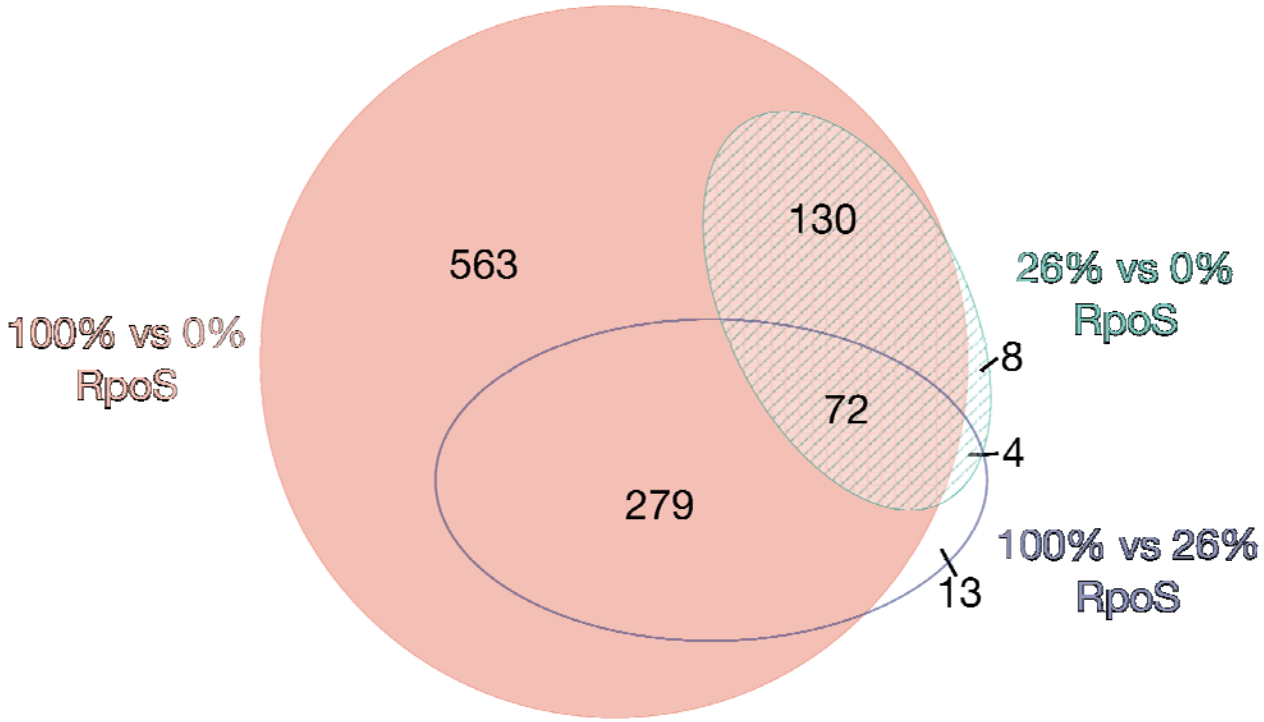

Figure 3: Area proportional Venn diagram showing the number of genes differentially expressed between each of the three conditions.

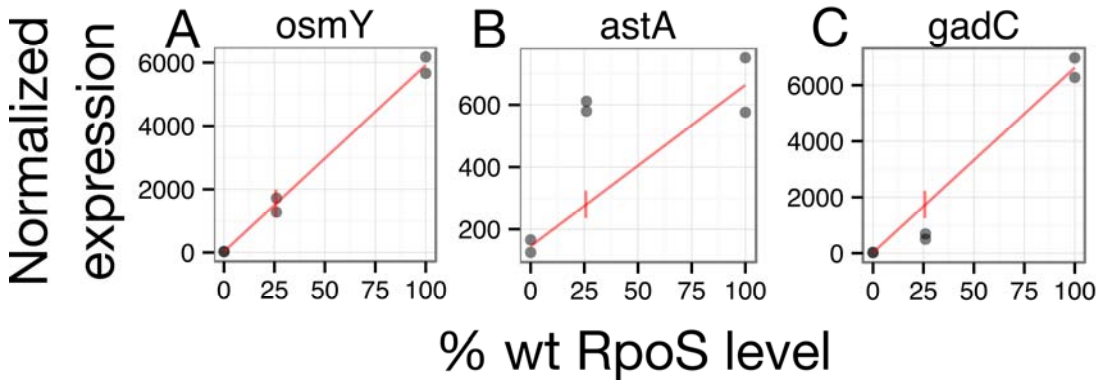

Figure 4: Examples of three classes of patterns of RpoS response. *osmY* expression is linear with RpoS level, *astA* is sensitive to RpoS level, and *gadC* is insensitive to RpoS level. Dots represent normalized expression levels from individual RNA-seq samples. The line is drawn between the mean at 0% and the mean at 100%.

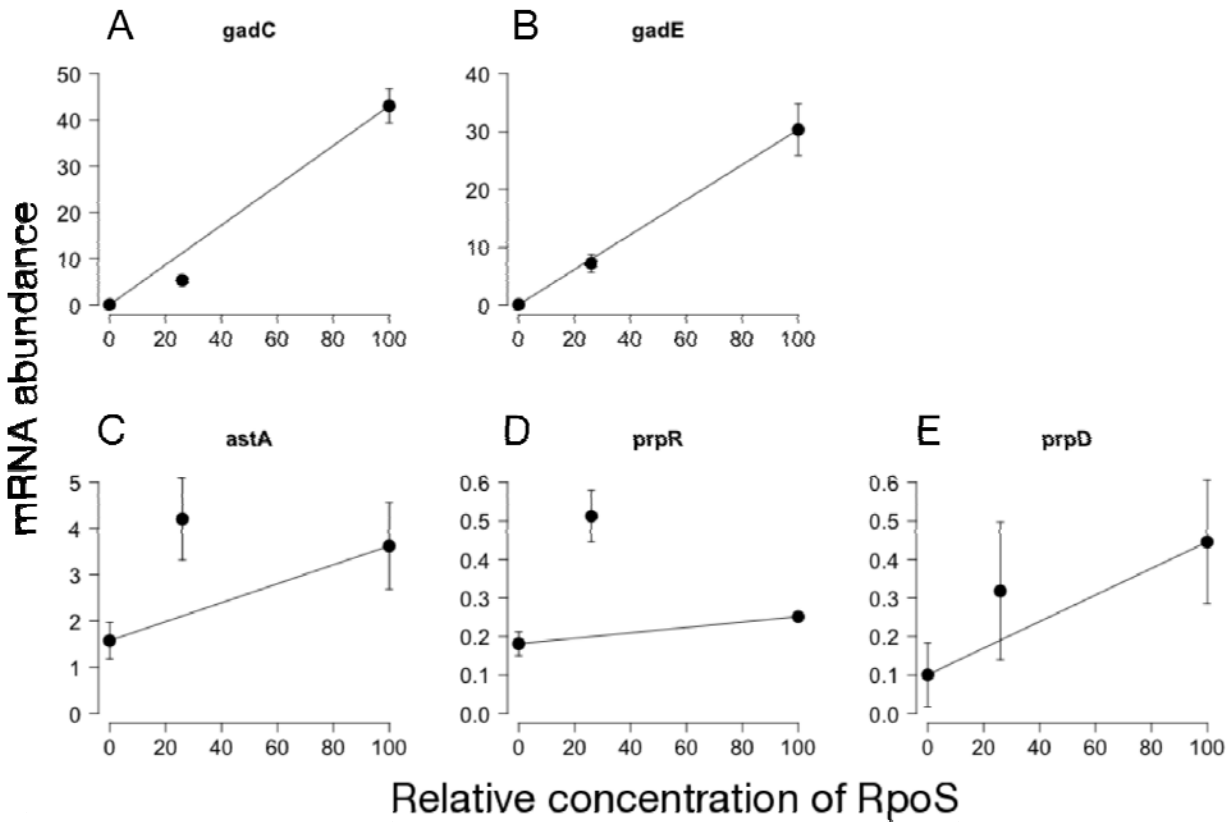

Figure 5. Testing of RNA-seq expression patterns using qPCR. Expression patterns were measured for (a,b) *gadC* and *gadE*, which were insensitive in the RNA-seq data, and (c-e) *astA*, *prpR,* and *prpD*, which were sensitive in the RNA-seq data. In all cases, the level of transcription at 26% RpoS was on the side of the line predicted by the RNA-seq data, though not always significantly so. *gadC* was significantly insensitive ($p < 10^{-4}$), *gadE* was not significantly different from linear ($p = 0.10$), *astA* was not quite significantly sensitive ($p = 0.06$), *prpR* was significant sensitive ($p < 10^{-4}$), and *prpD* was not significant ($p = 0.56$). n = 6, error bars = SEM.

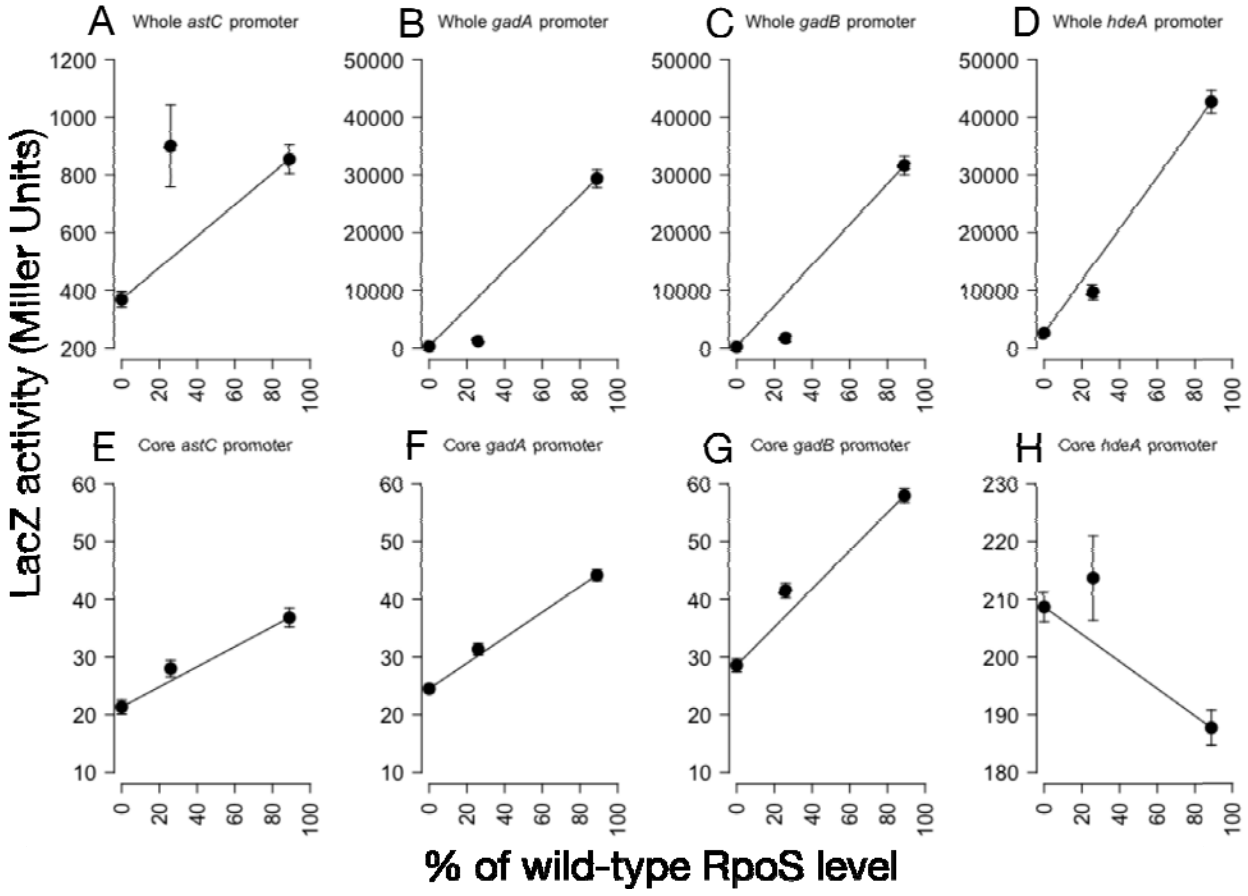

Figure 6. **Expression patterns of whole promoters and core promoters only.** Expression patterns were measured for the (a - d) whole upstream regulatory region and the (e - h) core promoter only of *astC, gadA, gadB,* and *hdeA.* As expected by RNA-seq data, the whole *astC* fusion was sensitive (p = 0.04, t-test), and the *gadA*, *gadB,* and *hdeA* fusions were all insensitive ($p = 2 \times 10^{-6}$, $p = 10^{-5}$, p = 0.04, all by one sample t-test). The p-values are adjusted for multiple comparisons using Holm's sequential adjustment method (51). The core promoters (e - h) have much lower levels of maximal transcription than the full length promoters. All four have much altered patterns of sensitivity, although only *gadA* and *gadB* was significantly different from the whole promoter (p = 0.08 (*astC*), p = 0.01 (*gadA*), $p = 10^{-6}$ (*gadB*), p = 0.21 (*hdeA*), all by two-sample t-test with Holm's correction.) n = 6 to 8, error bars = SEM.

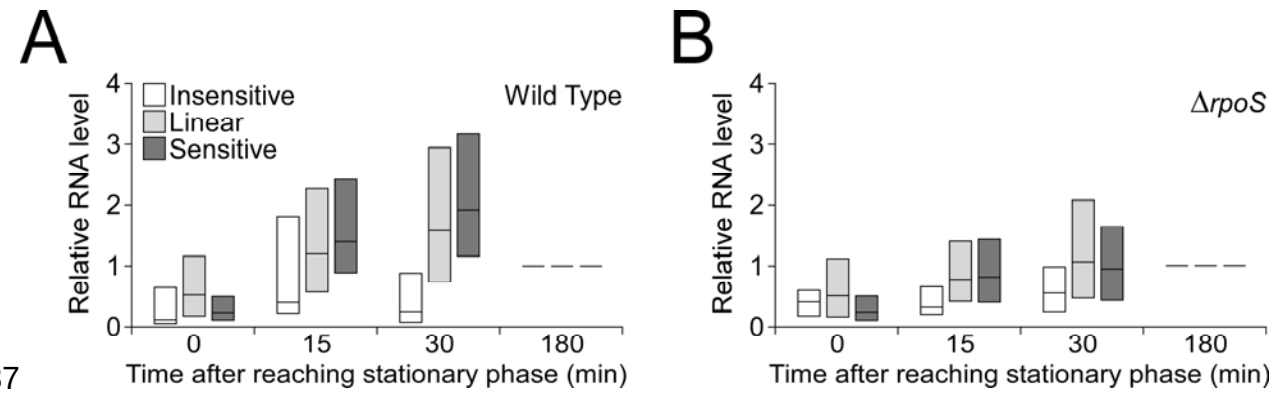

Figure 7. Expression profiles of genes when cells enter stationary phase. **(A)** Relative expression levels of RpoS-regulated genes were taken from a published study (48) for four time-points following entry into stationary phase. All expression values were normalized to those from the final time-point for each gene. The graph shows the range of relative expression values for insensitive (white boxes), linear (light gray boxes), and sensitive (dark gray boxes) genes for each time-point. Boxes represent the 25^{th}-75^{th} percentile range, and horizontal lines indicate the median value. **(B)** As above, but for a Δ*rpoS* strain of *E. coli*.